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THE PLANT DISEASE REPORTER

Issued By

THE PLANT DISEASE SURVEY

Division of Mycology and Disease Survey
BUREAU OF PLANT INDUSTRY, SOILS, AND AGRICULTURAL ENGINEERING
AGRICULTURAL RESEARCH ADMINISTRATION
UNITED STATES DEPARTMENT OF AGRICULTURE

Volume 32 Number 6

June 15, 1948

The Plant Disease Reporter is issued as a service to plant pathologists throughout the United States. It contains reports, summaries, observations, and comments submitted voluntarily by qualified observers. These reports often are in the form of suggestions, queries, and opinions, frequently purely tentative, offered for consideration or discussion rather than as matters of established fact. In accepting and publishing this material the Division of Mycology and Disease Survey serves merely as an informational clearing house. It does not assume responsibility for the subject matter.

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DIVISION OF MYCOLOGY AND DISEASE SURVEY

Plant Industry Station, Beltsville, Maryland
Volume 32, June 15, 1948, Number 6

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BACTERIAL RED XYLEM DISEASE OF POTATO TUBERS IN MAINE

Donald Folsom, J. S. Getchell, and Reiner Bonde

A reddish discoloration of the xylem occurs in potato tubers in Maine which is caused by bacteria other than those of the soft-rot or black-leg kind. Some reference was probably made by Bonde to cases of this disease in 1939 (1, Tables 1 and 2, Isolates M 15-17 and 28-30). Also, it was reported to be one of the major causes of graying of dehydrated potatoes in the 1944 crop in Maine (4). Potato seed and table-stock inspectors find it in Maine at digging time and in storage, where an associated symptom often is a corrosion of the tissue at the stolon scar, and have reported up to about one percent infection in a stock. Terminal market inspectors have been observed to call the disease Fusarium wilt discoloration although Fusarium wilt is absent in Maine.

Pathogenicity of culture can be tested by smearing broth cultures onto freshly cut tuber slices in fairly humid air in desiccators or in saturated air in petri dishes. In the former the results are dark brown to black lesions sometimes several cm. across and typically several mm. deep but sometimes extending into the tissue for several cm., and some xylem discoloration. In the petri dishes the result is the rapid development of a shallow yellow rot tinged more or less with black. The pathogen has been isolated from the stolon-scar lesions, from the red xylem to distances of 10 or more cm. from the stolon scar, from red streaks extending from the stolon scar into the central part of the tuber, and from lesions in the cortex of "pitted" tubers that apparently have been wet in storage. It has also been reisolated from the lesions and xylem of inoculated tissue and used for successful re-inoculations.

The surface of the stolon-scar lesion is depressed but not discolored, while its interior is dark brown to black and a cavity may develop. The color of the "red" xylem in Katahdin tubers is antimony yellow to ochraceous tawny by Ridgway (3, table XV) and matches the range of color shown by Maerz and Paul (2) in 10-H-5, 11-G-5, or 12-F-6, when exposed as a network by removing the cortex. Affected Katahdin tubers are usually comparatively large. The pathogen is generally replaced by various other kinds of bacteria or miscellaneous fungi or both during storage, so that by January in warm storage and by March or April in cold storage it may no longer be possible to isolate the organism. The consistent presence of the red xylem at the proximal or stem end of the tuber, the frequent subepidermal lesion at the stolon scar, and the presence of the disease at digging time, all indicate that the organism often enters the tuber from the parent plant. The large size of many affected tubers suggests that the parent plant may become affected early enough for some tubers to be cut off in time to influence the size

of the remaining tubers. The disappearance of the pathogen from the xylem during the storage period explains the inability of the writers to demonstrate perpetuation of the disease through infected seed tubers. The inoculation of plants through seed pieces or otherwise has not been feasible yet.

The red xylem has more or less resemblance to the vascular discoloration of tubers attributed to the use of herbicides, but the xylem in the latter case is sterile until well along in storage and even then is usually sterile. The color is different from that of stem-end browning and net necrosis, the latter affecting only the phloem. The Katahdin variety, immune to stem-end browning and net necrosis, is the one most commonly affected.

Preliminary studies of the pathogen show that on artificial media, the organisms occur as gram negative, motile, nonsporeforming, noncapsulated short rods (1.5 mu to 2.0 mu x 0.5 mu to 0.8 mu) single and in pairs. Colonies are white to translucent, circular, slightly raised, 1.5 to 2.0 mm in diameter; edges are lobate; growth is ringed. In liquid media, growth is at the surface with pellicle formation; sediment is slight; facultative to oxygen. On agar strokes, growth is filiform and moderate with slight iridescence in reflected light. Neither acid nor gas is produced from glucose or lactose; neither soluble starch nor raw potato starch is hydrolyzed; indol, hydrogen sulfide, and acetylmethyl-carbinol are not produced; nitrates are not reduced; gelatin is rapidly liquefied; litmus milk digested without coagulation. Growth on potato slants is moderate with dull orange pigment; potato blackened. Growth temperature ranged from 6°C to 37°C; optimum 25 to 30°C.

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UNIVERSITY OF MAINE
ORONO

PROGRESS REPORTS ON LATE BLIGHT, TOBACCO BLUE MOLD, AND CUCURBIT DOWNY MILDEW

(Summarized from reports of Key Pathologists of the Crop Plant Disease Forecasting Service received up to May 15)

LATE BLIGHT (*Phytophthora infestans*)

Potato late blight became increasingly active during the latter part of April in the FLORIDA Everglades section and was very destructive in untreated or carelessly treated fields. Digging was started at the beginning of May and yields were sharply curtailed where control programs were lax. Near Gainesville in north-central Florida infection, apparently a week or so old, was observed on blossoming potato plants at the end of April, but dry weather in that section at that time was unfavorable to spread of the disease.

After a period of quiescence or absence on tomatoes in Florida generally, late blight resumed activity on this crop during the last part of April in the Homestead and the Everglades areas. In both cases reappearance was correlated with a period of cooler weather, and in the Everglades with four successive rainy days as well. Near-epidemic attack occurred in some of the few remaining tomato fields in the Homestead section. It was thought that late blight had never previously been observed in that area so late in the season (date of observation, April 28). In the Everglades the present infection on tomato plants was described as not the same in aspect as during the winter months, with no stem lesions observed, fewer leaf lesions per plant but more rapid killing of attacked leaflets, and less sporulation.

In TEXAS continuously dry weather checked the potato late blight infection reported in the Lower Rio Grande Valley early in March (PDR 32 (4): 131. Apr. 15), and up to harvest time in the latter half of April no further infection was found on either potato or tomato, nor had the disease been reported on either host from other parts of the State.

After an unfavorable period in LOUISIANA, rainy cloudy weather on April 12 and 13, followed by cool nights with heavy dews, gave ideal conditions for late blight, and the disease spread rapidly on potatoes in Terrebonne Parish. It reached epidemic proportions in some fields, although relatively little occurred in sprayed or dusted plantings. Harvesting was in progress during the first week of May. Small infection centers were found on potatoes in Lafourche Parish. None has been found or reported on tomatoes in the State.

In MISSISSIPPI tomato late blight was found May 11 near Crystal Springs in one small well-isolated field planted with transplants obtained from Texas five or six weeks earlier. Cool nights and very heavy dews favored infection.

Potato late blight, apparently originating from diseased seed, was found in ALABAMA April 5, in Baldwin County, and was spreading rapidly by April 9. Copper dusts were the most widely used fungicides. A dithiocarbamate dust was being tried by a few growers. Dithane D-14 was being used by those who preferred sprays. However, many growers were doing little or nothing to control the disease.

In SOUTH CAROLINA, a few lesions of potato late blight were found in one field on Johns Island in Charleston County, April 16. No more was found at that time in nearby fields. A survey made on April 22 on Edisto Island, on the same farms where blight was first seen in 1946 and 1947, revealed lesions quite generally distributed over the area and at least one small spot where the plants were almost completely dead. On the same day the disease was reported from a farm near Charleston. The location of all these infections indicated that late blight was spread over the southern half of Charleston County, where most of the State's potatoes are grown, and was in a position to cause severe damage with the occurrence of a single wet cool period. However, no such weather developed and no visible advance took place during the remainder of the month, although lesions continued to show some spores each morning. Some spread occurred during a rainy period in the first week of May. The disease had not reached serious proportions, however. None has been found in Beaufort County.

On tomatoes, general light infection was found May 11 in a 30-acre field on Johns Island in Charleston County.

In VIRGINIA, tomato late blight was found in Northampton County, on the southern tip of the Eastern Shore, on April 27, on plants shipped from southern Georgia and set out about two weeks previously. About twelve acres had already been plowed up. The disease was difficult to diagnose at first since the plants were in poor condition, but cultures showed growth of the fungus. The grower had dusted with a copper dust since the plants were set out, but evidently the prior infection nullified the effect of the dusting.

TOBACCO BLUE MOLD (Peronospora tabacina)

Although spread was slow most untreated beds in SOUTH CAROLINA were affected a month after the first appearance of blue mold on March 15. Surveys showed infection just starting in most beds April 9, and most spread took place after a cool rainy period April 10 to 12. Since then warm weather allowed recovery and no serious delay in transplanting occurred. Most of the crop was being set out during the last two weeks in April. Apparently fewer growers used treatment this year, probably because of the light attack; about 4000 of 17000 yards of plant beds observed in two surveys were being treated. Striking contrasts were observed in several locations between large vigorous plants on treated beds and delayed infected plants on untreated beds. One county agent reported

that some growers deliberately allowed infection of their beds to hold back their plants, a hazardous practice if conditions should become favorable to the disease.

In NORTH CAROLINA, by the middle of April, blue mold was present over the eastern half of the State except in the northeast. Damage generally was light to moderate, with severe injury and killing of plants in a number of isolated cases. If weather remained warm no serious trouble was anticipated. Transplanting was being started in the southern counties. On some farms transplanting delay would be caused by blue mold, but at the time of the last report (April 17) most of it, especially in the old belt, would be due to frequent rains holding up land preparation. Fermate spray or dust was being widely used with good results where properly applied. At five locations in Lee County blue mold was observed to be more severe in beds on old sites and infection apparently had originated first in old bed sites. In Moore County a striking situation was noted: in a plant bed on one side of a creek where the Uramon-Cyanamid treatment had been used last fall, weed control was excellent and little blue mold was observed; whereas in a bed across the creek where no weed control treatment had been used, weeds were rampant and blue mold was severe.

In VIRGINIA blue mold has been reported in five southern counties, Halifax, Pittsylvania, Mecklenburg, Brunswick, and Nottoway, first reports being received about the middle of April, although some evidence indicated that the disease had been present in the region for ten days or so, perhaps as early as April 4 or 5. Warm dry weather prevented serious damage. Plants were in good condition and setting out was starting by the first week of May.

In MARYLAND blue mold has been reported in Anne Arundel, Calvert, Saint Marys, and Prince Georges Counties. Weather following first observations, on April 23 in Anne Arundel and Calvert Counties, was favorable to the disease, with cool temperatures and some rain or drizzle on most days. However, damage was slight up to the first part of May, the general use of Fermate apparently preventing serious attack.

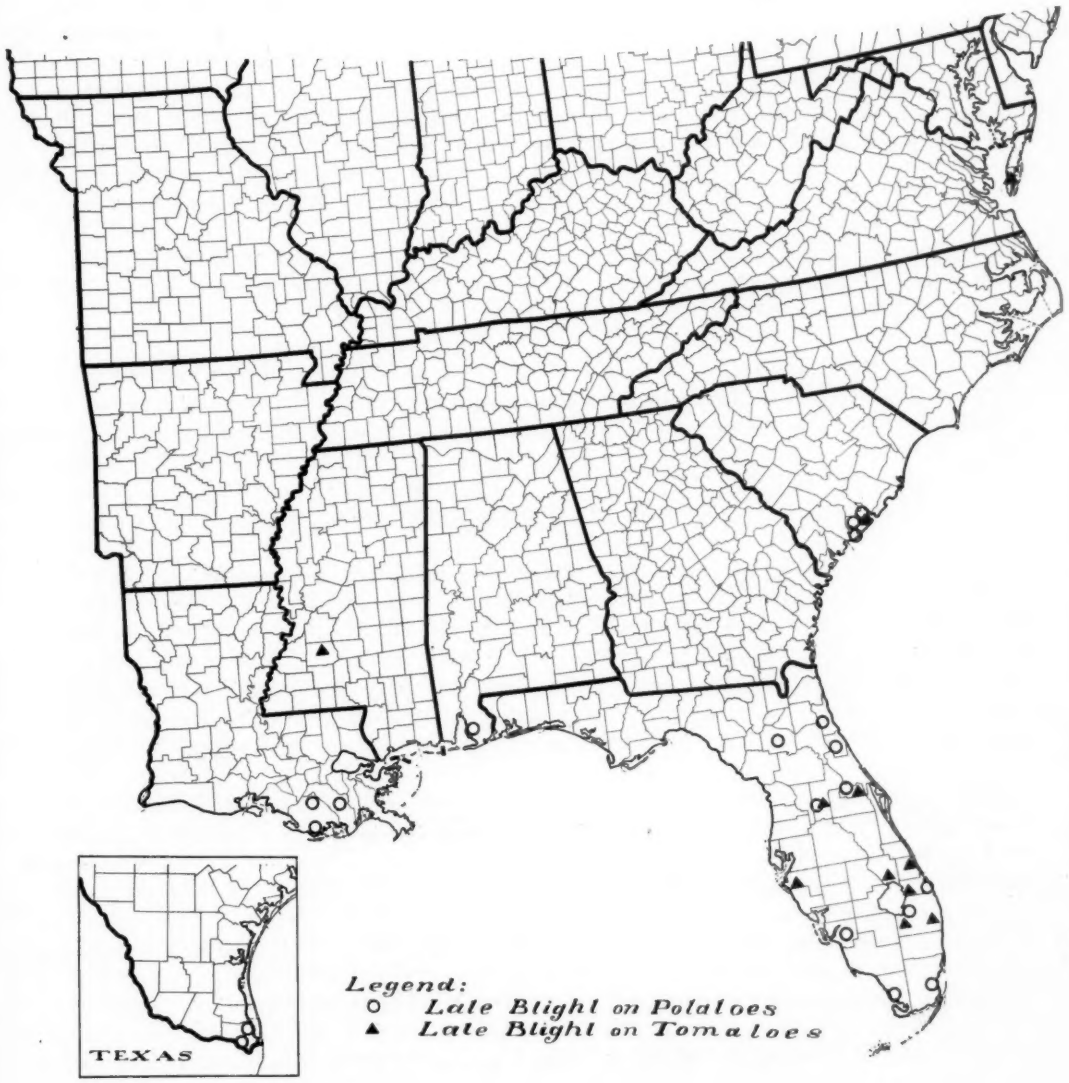
Blue mold was first found in TENNESSEE on April 30 in Green County. During the first week in May conditions were ideal for its development and it was widespread over the Burley area, but was generally being well controlled.

In KENTUCKY the disease was first found April 27 in Simpson County in second-year beds. Up to May 10 scattered infections were reported in several counties, but no serious injury was caused.

DOWNY MILDEW (Pseudoperonospora cubensis) OF CUCURBIT CROPS

By the middle of April the squash and cucumber season on the lower East

Fig 1. **LATE BLIGHT** in 1948:
occurrence reported in Warning Service Area up to May 15



Coast of FLORIDA was over, with only a few small plantings remaining. According to reports most of the squash and cucumber fields in the Fort Lauderdale-Pompano area were killed by the disease. In the Leesburg area the disease was found on watermelons April 15. By May 7 it was severe in some parts of the area and slight or absent in others. Temperatures during the preceding ten days ranged from 88° to 94° F in the daytime and dropped to the low 60's at night, with heavy dews. At the end of April commercial cucumber fields in the vicinity of Gainesville were affected, infection being widely distributed but scattered. Dry weather apparently had limited secondary spread from the isolated primary infection localities.

A grower in Colleton County, SOUTH CAROLINA, reported downy mildew on cucumbers May 11, stating that the disease was well-established but causing negligible damage. A second report was received from Charleston County May 12.

INCIDENCE OF LATE BLIGHT, TOBACCO BLUE MOLD, AND CUCURBIT DOWNY MILDEW IN THE FIRST PART OF 1948

The distribution of late blight on potato and tomato, and of tobacco downy mildew, as it was reported by Key Pathologists of the Crop Plant Disease Forecasting Service up to May 15, is shown on the accompanying maps (Figures 1 and 2).

LATE BLIGHT (*Phytophthora infestans*) ON POTATO

Late blight was already established on potatoes in the lower East Coast, Everglades, and West Coast areas in FLORIDA late in December or early in January. It became general and important, thorough spraying being necessary to control it. It caused serious loss in untreated or improperly treated fields in the Everglades spring crop. In the Hastings section, where it appeared early in March, thorough and prompt control prevented an epidemic.

Potato late blight appeared in the Lower Rio Grande Valley of TEXAS at the beginning of March but was promptly stopped by dry weather.

In southern **LOUISIANA** the disease appeared on the fall crop in Terrebonne Parish early in December and many of the tubers from that crop were badly affected when dug in January, which is the first record of late blight in tubers from the fall potato crop in that State. Unfavorable weather prevented digging of the crop for several weeks after the plants were severely affected. On the spring crop in Terrebonne Parish infection appeared at the end of March but made little headway until more favorable weather in mid-April, when it became epidemic in untreated or poorly treated fields. A little blight appeared in

Lafourche Parish also.

Infection apparently originating from diseased seed appeared in Baldwin County, ALABAMA, early in April and spread rapidly. Effective control measures were being used by some growers but many were making no serious attempts at control.

The disease was found in three locations in Charleston County, SOUTH CAROLINA, about the middle of April or shortly after, the distribution indicating that the southern half of the County, where most of the State's potatoes are grown, was widely affected. Weather was mostly unfavorable for spread until a rainy period during the first week in May when some took place.

LATE BLIGHT ON TOMATO

As with potatoes, late blight appeared on tomatoes throughout the southern half of FLORIDA in December or January. It caused serious loss to the fall crop in the Homestead and other East Coast areas, and generally required thorough spraying for control. On the spring crops the disease was generally less active and by the end of March it had ceased to be a factor even where not controlled. However, it revived in the Everglades and Homestead areas with cooler weather during the latter part of April. Infection of seed-beds for transplants was also evident throughout the period, amount varying with weather but always present.

Outside of Florida, tomato late blight so far has been reported only from MISSISSIPPI, in one small field near Crystal Springs, May 11; SOUTH CAROLINA, in one field in Charleston County, May 11; and VIRGINIA, in Northampton County on the Eastern Shore, April 27, on plants shipped from southern Georgia and set out about two weeks previously.

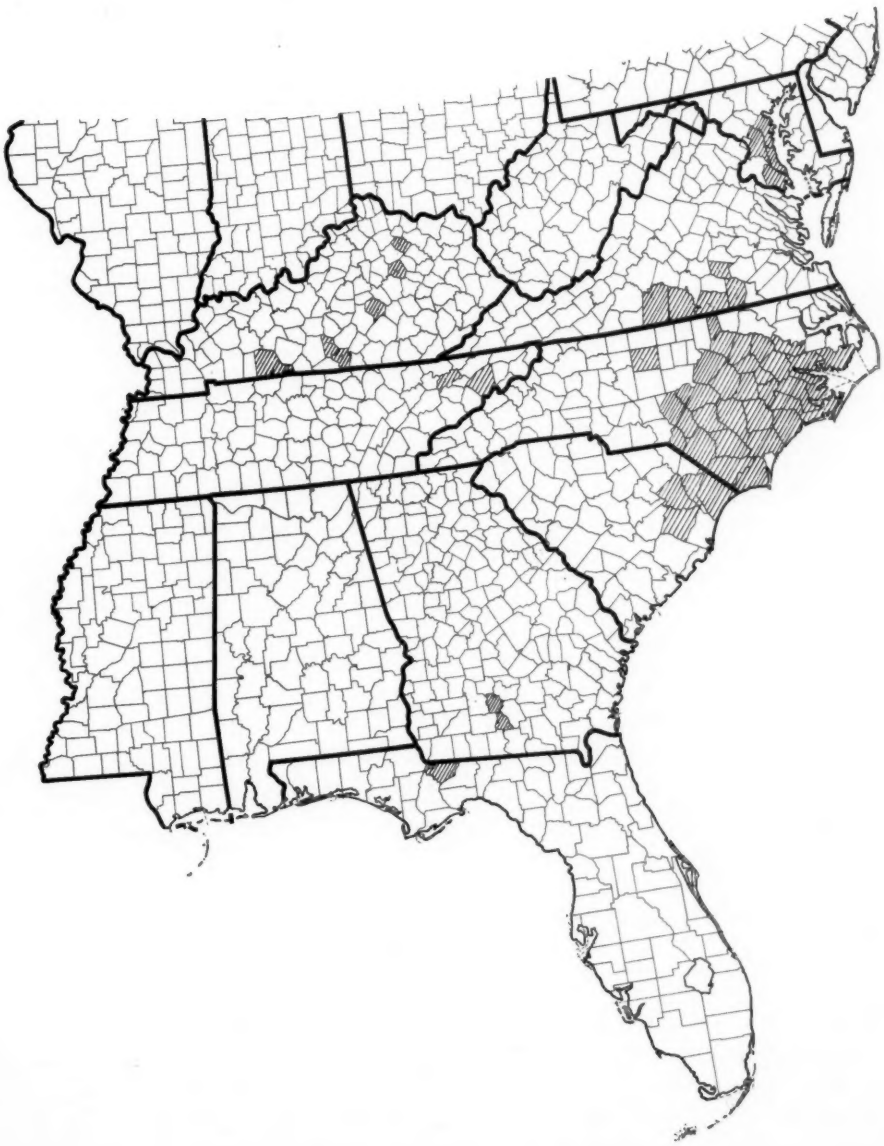
TOBACCO BLUE MOLD (Peronospora tabacina)

Blue mold was reported from Cook County, GEORGIA, as early as January 28. Since then it has appeared as far north as Maryland, and in Kentucky and Tennessee, being first reported in FLORIDA February 23, in SOUTH CAROLINA March 15, in NORTH CAROLINA March 23, in VIRGINIA April 17 although probably occurring a week or so earlier, in MARYLAND April 23, and in KENTUCKY and TENNESSEE at the end of April. Fermate spray or dust was widely used. So far the disease has not been severe and has caused no delay in States where plants are being set out.

CUCURBIT DOWNY MILDEW (Pseudoperonospora cubensis)

So far this disease has been reported only from FLORIDA and from SOUTH CAROLINA. It had caused some local loss in the former State, especially on cucumbers, and was just appearing on watermelons there. In South Carolina it was reported on May 11, on cucumbers in Colleton County.

Fig 2. TOBACCO BLUE MOLD in 1948:
occurrence reported in Warning Service Area up to May 15



ASTER YELLOWS IN CARROTS IN IDAHO

R. D. Watson

In the period from 1943 to 1945, in southwestern Idaho, the aster yellows virus disease in carrots grown for seed developed until it became of major importance. From 30 to 50 percent of the carrot plants in some fields were infected. Its occurrence is correlated with the rapid expansion of the acreage in that area, and it may have come in with carrot roots shipped into Idaho. The aster yellows virus never became well established in the weed suscepts. It was reported as occurring only on those weeds which were near heavily infested fields. It was present chiefly in the carrot crop, infecting the steckling from the seed crop. The stecklings were usually grown in the same field with the seed crop.

A control program was initiated by the University, with seedsmen cooperating, to break up the cycle of diseases from seed crop to steckling of both aster yellows and bacterial blight by planting stecklings isolated from the seed crops. No accurate measure is available to indicate how much these control measures of isolating the carrot steckling planting from the seed crop had on the control of aster yellows disease, but the rapid decline in the importance of the diseases seems significant and may be due partly to the control measures applied on 1944 and succeeding crops. In 1945 8 to 12 percent of the carrot seed plants were infected with aster yellows, indicating infection in 1944 stecklings. In 1946 and 1947 no current season symptoms were observed either in carrot or lettuce fields. In carrots only a trace of aster yellows was found in most seed fields with the highest counts showing 2 percent infection in 1946 and none in 1947.

This is an interesting case of a disease new to Idaho becoming serious and then apparently dropping out of the picture for two years at least. Aster yellows continues to be an important disease in certain areas in nearby States, with ample opportunity for the reintroduction of both the virus and its insect vectors into southern Idaho.

UNIVERSITY OF IDAHO

CARROT BACTERIAL BLIGHT CONTROL IN IDAHO

R. D. Watson

Carrot bacterial blight (Xanthomonas carotae) probably has been present in Idaho since 1941, and from 1942 to 1945 it has been a disease of major importance. In 1944 a cooperative control program was established between the seed companies and the University of Idaho[1]. A hot water seed treater was developed by the University and used by the seed companies to treat their seed planted in the summer of 1945[2,3].

The first major field test for the control measures was available in 1946. Seed was treated in 1945 and planted that summer. The stecklings were stored and planted for the seed crop in 1946. In order to evaluate the degree of control obtained, a survey was conducted June 27 to 29 and August 6 to 10, 1946.

The control measures used were: (1) the seed was treated with hot water to kill bacteria in the seed, (2) the stecklings were planted in a field isolated from the seed crops, (3) the stecklings and seed crop were planted on ground new to carrots, (4) the seed fields were isolated from other carrots.

For a good test to prove the value of these control methods, it would have been necessary to plant untreated seed and avoid isolations; but all concerned were convinced of the value of the measures and were interested only in cleaning up the disease in the carrot seed crop in Idaho. For this reason almost all of the companies concerned treated the seed involved and left no good check available in southern Idaho for comparison. In two cases growers reset stecklings on fields that had carrot seed on the year before, giving some evidence of the value of control methods. This is further proof of the carry-over of the pathogens in the soil, since in both cases some of the same lot of stecklings planted on new ground had no bacterial blight.

Twenty-eight carrot seed fields were chosen to be inspected, representing a large percentage of the acreage and seed lots grown in Idaho.

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[1]Seed companies supplying data used in the report are Associated Seed Growers, Inc., Nampa, Idaho; Crockham Company, Caldwell, Idaho; Pieters-Wheeler Seed Co., Caldwell, Idaho; and Ferry-Morse Seed Co., Caldwell, Idaho.

[2]Blodgett, Earle C., and Glen KenKnight. A practical hot water seed treater. Seed World. Oct. 19, 1945

[3]Ark, P. A., and M. W. Gardner. Carrot bacterial blight as it affects the roots. Phytopath. 34: 416-420. 1944

All were grown from hot water-treated seed. Thirteen fields had no blight present. Of the thirteen fields with blight present in traces, only one infected head was found in six of them. Two fields planted on soil having seed crop the previous year had 15 percent and 60 percent of the heads infected.

There was evidence that the bacteria are carried to flowers by insects. Systemically infected plants and leaf symptoms were commonly found in the two fields planted on old carrot ground but not in the other fields. Blight was known to have been present in one of these fields the preceding year, and in this one 60 percent of the plants had infected heads. This field (No. 1) is compared with other fields in the same area as to the amount of blight, seed germination, and yield per acrea.

Field No.	Bacterial Blight	Germination Percent	Yield per acre Pounds
1	60% heads infected	69	490
2	None	92	970
3	None	88	930
4	None	85	880

The 1946 and 1947 survey of seed fields grown from hot water-treated seed and planted in isolation from carrots and on ground new to carrots clearly demonstrated the practical value of these control measures in controlling bacterial blight on a large scale field test. If these control measures are followed carefully by the seed companies, then the carrot seed produced in Idaho will likely remain free of the bacterial blight organism and free from the loss in germination and yield due to the disease. Losses in yield due to bacterial blight can be completely eliminated in Idaho, as demonstrated by this field scale cooperative test of the control measures.

UNIVERSITY OF IDAHO

WHITE AND GRAY MOLDS ON BLUE LAKE BEANS IN WESTERN OREGON DURING 1947

Frank P. McWhorter

During 1947 the Oregon Experiment Station initiated control experiments to protect Blue Lake beans from white mold, Sclerotinia sclerotiorum. Since the results of these investigations are contrary to the statements concerning the occurrence of this disease in Oregon made by Zaumeyer and Thomas, (PDR 31: 441, 1947) the present comments have been prepared.

The Sclerotinia disease of beans has been studied in Oregon intermittently since 1916, when Elmer[1] plotted the abundance of apothecia with reference to the time of the year. It was shown that they are most abundant during late fall and early winter, scarce during late spring and very scarce during the summer months. A more recent study of the apothecial distribution verifies the above data; however, they were very abundant during February and March of the 1946-47 season. In addition to the natural reduction in development of apothecia as summer approaches, the mode of cultivation of bean fields reduces the number of sclerotia on or near the surface of the soil, thus reducing the possible importance of ascospores as initial means of infection. Contrary to the statements of Zaumeyer and Thomas, at least 90 percent of the initial infection of Blue Lake beans occurred at the soil line where there was direct contact of the plants with phases of the fungus in the soil. The disease has become more important during the past three years. One probable (but unproved) reason for this is the fact that the new "stringless strains" of Blue Lake beans have a more "leafy" habit of growth. The plants appear spread out at the bases, more leaves touch the ground, thus providing an excellent inoculation court for the fungus at the soil line.

A good growth of Blue Lake beans develops compact rows of vines eight feet high. Since there is no practical means of removing this mass of vines from the field at the end of the picking season, the practice is to leave the vines on the ground to rot. In areas where Sclerotinia infection is present, the fallen vines already contain sclerotia. As rot progresses, the decaying vines serve as a culture medium for the fungus, enabling it to form new mycelial masses and countless small sclerotia. The vines are in an advanced state of rot by January. The bean fields are consistently placed in low locations. Unfortunately the breaking up of the vines from rot is coincident with spring floods which cover the bean fields to a depth of several feet. Flood waters are an effective means of transporting bits of bean stems bearing sclerotia intra- and inter-fields. When the fields become dry enough in the

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[1] Elmer, O. H. Sclerotinia rots of Leguminous plants. Unpublished Master's Thesis, Oregon State College Library.

spring all the remaining debris is thoroughly cultivated into the soil. This practice largely circumvents further apothecial development but it favors soil infestation.

The possibility of controlling the disease by applications of 500 to 1500 pounds of calcium cyanamid was studied in trial plots and commercial plantings. These applications were made to the soil and remaining plant debris during the early spring months when the apothecia were abundant. At the time of application there were from 10 to 26 apothecia per square foot of test row. Apothecia touched by the chemical withered shortly after it was applied. Later the soil was thoroughly cultivated and beans were planted. Infection counts made on comparable treated and untreated rows showed no significant reduction in infection. Each developed approximately 20 infection centers per 100 feet of test row. There were no apothecia found in the heavily treated rows.

In order to explain the blossom infection and rapid spread reported by Zaumeyer and Thomas, it is necessary to quote these authors.

> White Mold. This disease was very serious in many of the Blue Lake fields. In others, smaller amounts of infection were observed. In many of the more seriously infected fields, 4 to 5 tons of beans were picked before they were abandoned, which is about 40 to 50 percent of a normal crop. It appeared that most of the infection resulted from ascospores. In many cases leaf infection started where dead plant parts, such as old blossoms, lodged on a leaf. Here the ascospores germinated and the mycelium proceeded to develop into the growing tissue. Rapid spread was noted, as conditions were ideal for the development of the organism since fields are watered with the overhead system of irrigation. Apothecia were not commonly observed but were found. The disease is increasing in importance and is becoming a very serious factor in production in these counties.

Unfortunately the observers did not see these fields earlier in the season when the presence of another fungus, Botrytis, on the "dead plant parts, such as old blossoms" could be readily discerned. During the unusual rainy weather of June, damage from Botrytis was more conspicuous than damage from Sclerotinia. Cultures from withering flowers consistently yielded Botrytis cinerea. When infected flowers fell on leaves Botrytis rot developed quickly at the point of contact. Thus flowers infected by aerially borne Botrytis spores materially reduced the yield of beans in Western Oregon in 1947. There is no available specific proof that ascospores from the very rare apothecia hidden under the leaves, were important in the initiation or progress of infection in Blue Lake plantings. On the contrary, trial inoculations with ascospores on Blue Lake foliage produced a different type of initial infection from that observed in the field.

OREGON STATE EXPERIMENT STATION

OCCURRENCE OF ANTHER MOLD IN LADINO CLOVER IN OREGON[1]

John R. Hardison

During the winter of 1946-47 abnormal seeds of ladino clover, Trifolium repens L. var. Ladino from southern Oregon that failed to germinate were submitted to the writer for examination by Mrs. L. A. Kanipe[2]. Such seed fails to germinate but is pervious to water, becomes greatly swollen and eventually is attacked by a variety of bacteria and fungi. However, on a few of the seeds a trace of a small, delicate Botrytis was observed, which appears to be identical with Botrytis anthophila Bond.

A few disinfected seeds have yielded a fungus which in culture appears identical with the characters described for B. anthophila by Silow[3].

In August 1947, one ladino clover flower was found near Grants Pass, Oregon which contains the typical "anther mold" stage of the disease. This specimen, however, was the only one found in several thousand flowers examined.

Recently, shrunken and shriveled seeds have been selected from various samples of 1946 ladino clover seed. From 750 disinfected seeds planted on potato-dextrose agar less than half of one-percent recovery of B. anthophila has been obtained. From all isolations to date there has been a 2.8 percent recovery of the fungus.

Apparently the seed generally must be damaged before this systemic fungus can be isolated from infected seeds on culture media. At present, the abnormal seeds described above cannot be correlated with B. anthophila, and the causes of such damaged seed are now being studied.

This appears to be the first record of clover anther mold caused by Botrytis anthophila in the United States, and also is apparently the first record of the disease anywhere on ladino clover. Occurrence of the disease on red clovers of American origin was indicated by Silow.

U. S. BUREAU OF PLANT INDUSTRY, SOILS, AND AGRICULTURAL ENGINEERING, DIVISION OF FORAGE CROPS AND DISEASES

1 Cooperative studies between Oregon Agricultural Experiment Station, Department of Plant Pathology, and U. S. Department of Agriculture Bureau of Plant Industry, Soils, and Agricultural Engineering, Division Forage Crops and Diseases at Corvallis, Oregon.

2 Seed Analyst in charge of the Cooperative Federal-State Seed Laboratory, Corvallis, Oregon.

3 Silow, R. A. A systemic disease of Red Clover caused by Botrytis anthophila Bond. Trans. Brit. Mycol. Soc. Vol. 18: 239-248. 1933-34.

FURTHER OBSERVATIONS ON BACTERIAL BLIGHT OF ORCHARD GRASS IN OREGON[1]

John R. Hardison

In addition to the infected plants of orchard grass, Dactylis glomerata L., found in May 1945[2], bacterial blight (Rathay's disease), Corynebacterium rathayi (E.F.Sm.) Dowson, was found in May and June 1946, and 1947 in the old observational row nursery of the U. S. Department of Agriculture on the Oregon Agricultural Experiment Station at Corvallis, Oregon. Infection was noted in a wide variety of orchard grass collections. Observations suggest that the disease probably has been present in these plantings for several years, possibly from the time the nursery was started, 1935.

Typical symptoms of severe stunting were observed for the first time in 1946. Prevalence of the disease in these old plantings varied from none to moderate, with severity ranging from slight infection in the panicles to severe stunting involving all above ground parts of the plant.

It appears that the infestation in the old nursery may have started from perhaps two or three infested seed lots planted in the triplicate rod rows. The spread appears to have been primarily to adjacent rows. There is very little suggestion of spread by cultivation equipment. It is remarkable that the disease appears to have spread laterally for only a few rows, and in most cases within affected rows only a few plants were severely attacked. Even in the most severely diseased rows fewer plants are affected than might have been expected from an outbreak of this disease.

In the U. S. Department of Agriculture uniform grass nursery the disease was observed again in Akoroa orchard grass. The prevalence was approximately the same as in 1945, but the severe stunting was observed, and in general the symptoms were much more severe than in 1945. A trace of infection was noted in an adjacent row of orchard grass, F. C. 22,763.[3]

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[1] Cooperative studies between Division of Forage Crops and Diseases, Bureau of Plant Industry, Soils, and Agricultural Engineering, United States Department of Agriculture, and Oregon Agricultural Experiment Station (Department of Plant Pathology), Corvallis, Oregon.

[2] Hardison, John R. Bacterial blight of orchard grass observed in Oregon. (Pl. Dis. Reptr. 29 (22): 600, 1945).

[3] F. C. numbers are accession numbers of Division of Forage Crops and Diseases, U. S. Department of Agriculture.

In the older nursery the disease was recorded on the following strains and collections of orchard grass:

Collection or Strain	Amount of Disease 1946	1947
Akoroa	trace	light (severe stunting)
Wash. No. 3135	trace	very light (severe stunting)
New Zealand Certified	light	very light
Sval of Skandia II	moderate	very light
Swedish F. C. 22366	moderate	very light
F. C. 22367	light	very light (severe stunting)
F. C. 22369	moderate	very light (severe stunting)
F. C. 22440	light (severe stunting)	trace
F. C. 22439 (Roskilde)	light	very light
F. C. 22438	light	trace
F. C. 22437	very light	none seen
F. C. 22370	none seen	trace (mod. stunting)
F. C. 22371	none seen	trace (in heads)
F. C. 22405	none seen	very light (in heads)
F. C. 22409	light	very light
F. C. 29580	none seen	trace

The disease has not been found in field plantings, and special attention has been given to the few field plantings of the Akoroa strain in Oregon.

U. S. BUREAU OF PLANT INDUSTRY, SOILS, AND AGRICULTURAL ENGINEERING, DIVISION OF FORAGE CROPS AND DISEASES

ADDITIONAL PARASITIC FUNGI ON GRAMINEAE IN THE INLAND EMPIRE[1]

Roderick Sprague and Jack P. Meiners[2]

The fungi listed herein represent additions to the check list being compiled by Sprague and G. W. Fischer on western grass and cereal diseases. We were aided in these additions by Conley V. Lowther, Wm. S. McGuire, and Ferdinand Wetter, Jr., who collected and studied mid-winter pink snow mold and leafspot material from Pullman and adjacent Idaho (Wetter) in class. The early winter in the Palouse region of Washington was relatively open with lush growth of pasture grasses and fall-sown grain. While January was less favorable for forage growth, there were a number of leaf spots, molds, blights, and mildews present. In January and early February Erysiphe graminis, Puccinia rubigo-vera, P. crandallii, P. poae-sudeticae, Rhynchosporium secalis, Scolecotrichum graminis, and Fusarium nivale were all present and the last two were prevalent.

The weather after February became less open with frequent snows and cold rains. Pink snow mold (F. nivale) became prevalent in the grass plots (Meiners) with some species showing considerable injury. We took general notes on severity, rating them from healthy (0) to dead from mold (10) but at this time only the additions to the check list are included. There was extensive pink snow mold damage in the winter wheat, especially in the wetter portions of fields. The total precipitation from September 1 to present writing (April 20) is more than 23 inches and the soils are heavy with excess water. What the farmers call winter injury appears to be partly due to F. nivale. The wheat died in very early spring, after the worst of the winter had passed. Certainly pink snow mold is an important disease in the Palouse. Blodgett[3] has reported similar observations from Idaho.

In addition to the winter collections and the pink snow mold notes, one of us (Sprague) collected material April 5-9 in the Cow Creek area of the Snake River Canyon in extreme N.E. Oregon. This interesting region can only be reached by power boat from Lewiston, Idaho, and is

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[1] Cooperative investigations between the Division of Plant Pathology, Agricultural Experiment Stations, Washington State College, and the Division of Forage Crops and Diseases, Bureau of Plant Industry, Soils, and Agricultural Engineering. U. S. Department of Agriculture. Published with the approval of the Director of the Washington Experiment Station.

[2] Associate pathologist, Division of Plant Pathology, Washington State College and Agent, Division of Forage Crops and Diseases, U. S. Department of Agriculture, respectively.

[3] Blodgett, E. C. 1946. Winter injury of fall seeded wheat in Idaho. Plant Dis. Reptr. 30(4): 106-111, 1 map.

a precipitous, semi-arid, mountainous, grassland area. Pink snow mold was present in mild form on Poa, Festuca, and Agropyron species while stripe rust (P. glumarum) and powdery mildew (E. graminis) were well advanced. The phenological development of vegetation is indicated by the fact that such plants as Amelanchier alnifolia, Dodecatheon sp., Poa cusickii, Arabis microphylla and Ranunculus glaberrimus were in full bloom.

The new state-host records, which are preceded by an asterisk are apparently new western records as well.

Fusarium nivale: occurs on the following additional hosts: WASHINGTON: *Agropyron dasystachyum (Hook.) Scribn.; *A. desertorum (Fisch.) Schult.; *A. michnoi Roshev; A. semicostatum (Steud.) Nees; *A. spicatum (Pursh) Scribn. and Sm.; A. subsecundum (Lk.) Hitchc.; *A. trachycaulum (Lk.) Malte; A. trichophorum (Lk.) Richt.; Agrostis alba L.; *Arrhenatherum elatius (L.) Beauv.; *Alopecurus arundinaceus Poir.; *A. pratensis L.; *Bromus arvensis L.; *B. brizaeformis L.; *B. catharticus Vahl; B. commutatus Schrad.; *B. erectus Huds.; *B. hordeaceus L. (European seed); *B. purgans L.; *B. rubens L.; *B. secalinus L.; *B. squarrosus L.; B. tectorum L.; *Cynodon dactylon (L.) Pers.; Dactylis glomerata L.; *Elymus canadensis L.; *E. sibiricus L.; *Festuca idahoensis Elmer; F. megalura L.; *Hystrix patula Moench; Holcus lanatus L.; *Hordeum brevisubulatum (Trin.) Lk.; *H. bulbosum L.; *H. jubatum var. caespitosum (Scribn.) Hitchc.; H. jubatum L.; *Lolium perenne L.; *Poa palustris L.; *Phleum phleoides (L.) Karst.; *Secale cereale x S. montanum; *Sitanion hystrix (Nutt.) J. G. Sm.; OREGON: Agropyron inerme (Scribn. and Sm.) Rydb.; A. spicatum; *Bromus rigidus Roth; B. tectorum L.; Festuca idahoensis Elmer; Koeleria cristata (L.) Pers.; Poa ampla Merr.; Poa cusickii Vasey; P. leptocoma Trin.; P. secunda Presl.; IDAHO: Agropyron intermedium (Host.) Beauv.; A. repens (L.) Beauv.; A. sibiricum (Willd.) Beauv.; A. subsecundum; A. trachycaulum; A. trichophorum; Bromus carinatus (Hook.) Arn; B. inermis Leyss; B. mollis L.; Dactylis glomerata; *Festuca elatior L.; *F. elatior var. arundinacea (Schreb.) Wimm; F. idahoensis Elmer (in Snake River Canyon above Lewiston); F. ovina L.; F. rubra L.; F. rubra var. commutata Gaud.; Poa ampla Merr.; P. bulbosa L.; P. pratensis.

In addition to the above listed citations, several other fungi have been found on new hosts in the several states:

Ascochyta agropyrina (Fairman) Trotter on *Agropyron spicatum, Pullman, Wash.-Ascochyta graminicola Sacc. var. brachypodii Trail on Poa cusickii Vasey, Cow Creek, Oregon.-Claviceps purpurea (Fr.) Tul. on Agropyron desertorum (old heads), Pullman, Wash.-Erysiphe graminis DC. on *Poa leptocoma Trin., Cow Creek, Oregon.-Fusarium acuminatum Ell. and Ev. on Agropyron michnoi, Pullman, Wash.-F. culmorum on Agropyron inerme, Pullman, Wash.-Pseudodiscosia avenae Sprague and A. G. Johnson on Arrhenatherum elatius, Moscow, Idaho-Rhizoctonia solani Kuehn, on

Agropyron subsecundum, Moscow, Idaho-R. solani on Festuca ovina L., Moscow, Idaho-Selenophoma donacis var. stomaticola (Bauemler) Sprague and A. G. Johnson on Poa cusickii, Cow Creek, Oreg.-R. solani on F. rubra var. commutata, Moscow, Idaho-S. donacis var. stomaticola on P. ampla, Moscow, Idaho-S. donacis var. stomaticola on Triticum aestivum, Union Flats, Wash.-Septoria bromi Sacc. on Bromus inermis, Moscow, Idaho-Septoria elymi Ell. and Ev. on Agropyron spicatum, Cow Creek, Oreg.-S. macropoda var. grandis Sprague on Poa cusickii, Cow Creek, Oreg.

WASHINGTON STATE COLLEGE AND U. S. BUREAU OF PLANT INDUSTRY, SOILS, AND AGRICULTURAL ENGINEERING

SENSITIVITY OF PENICILLIUM DIGITATUM TO 2,4-DICHLOROPHENOXYACETIC ACID

J. Guiscafre-Arrillaga

While testing various chemicals as possible inhibitors of postharvest decay of citrus fruit, a proprietary brand of 2,4-D (Dow 70), containing 70 percent of the acid and 30 percent $NaHCO_3$, was included in one of the trials. A marked decrease in decay occurred in oranges dipped for periods of 3 to 5 minutes in 0.1 to 0.5 percent solutions of this formulation. In subsequent tests similar results were obtained with another brand of 2,4-D (DuPont) containing 75.9 percent acid equivalent of the sodium salt when the same dilutions were used.

Rotting of non-treated fruit ranged from 14 to 25 percent in one week and over 50 percent in three weeks; fruit dipped in 0.1 percent solution showed 0 to 8 percent decay in one week; fruit dipped in 0.2 percent to 0.3 percent solution showed no decay in one week and an average of 10 percent decay in three weeks.

In other tests, instead of dipping the fruit in solutionsof 2,4-D, the chemicals were placed at the bottom of the container (1/2 bushel crate). Approximately 10 grams of the dry material, Dow 70, was scattered at the bottom of the container, covered with a layer of filter paper, and the fruit placed over it. An ethyl ester of 2,4-D (35.2 percent acid equivalent) was also used. Ten cc. of the undiluted ester were soaked on filter paper which was placed at the bottom of the container and the fruit packed over it. Marked reduction in fruit decay was obtained by these treatments. No rotting occurred the first week, with an average rot of 16 percent in three weeks, compared with over 50 percent rot in the check during the same period. While a faint odor of the 2,4-D was perceptible on the rind, the taste of the interior was in no way affected. Apparently the 2,4-D was absorbed by the outer layers of the rind.

Since the most prevalent type of decay in these tests was that caused by the green mold, Penicillium digitatum, it was decided to determine if 2,4-D would inhibit the germination of the conidia of this fungus. The following procedure was used.

A spore suspension was obtained by pouring 10 cc. of sterile distilled water on a 14-day-old slant culture of the fungus and rubbing the surface lightly with a rubber rod. One cc. of this heavy spore suspension was placed in 17 cc. of water, to which 2 cc. of filtered and centrifuged orange juice had been added. Spores did not germinate well in distilled water without the addition of orange juice. Two slightly different methods were employed to bring the spore suspension in contact with the material to be tested. In method 1, the desired dilution of the chemical

was smeared on glass slides, and, after drying, the spore suspension was placed on the dried smear. In method 2, a drop of the desired solution (or suspension) of the chemical was placed on the slide, a drop of the spore suspension placed next to it, and the two drops were then connected by drawing one into the other. Both methods gave comparable results. The slides were placed in moist chambers, and spore germination counts were made 24 hours later. The results of some of these tests, which are shown in Table 1, indicate that 2,4-D, in the concentration used, has a strong inhibitory action on the germination of conidia of P. digitatum. It has not been determined whether the action of 2,4-D on this organism is fungicidal or merely fungistatic, and the material was not tested against other fungi.

Table 1. Inhibition of germination of conidia of Penicillium digitatum.

Material and Concentration	Method 1 Percent germination			Method 2 Percent germination		
	Exp. 1	Exp. 2	Exp. 3	Exp.1	Exp.2	Exp.3
0.3% Dow 2,4-D[a]	0	0	2	0	0	0
0.2% " "	20	2	3	17	3	3
0.1% " "	13	8	14	11	11	9
0.3% DuPont 2,4-D[b]	1	0	1	1	0	0
0.2% " "	1	3	3	0	0	1
0.1% " "	5	9	9	7	9	9
0.04% $NaHCO_3$	81	86	91	97	91	93
0.06% "	91	89	89	95	95	89
Standard Bordeaux	0	0	0	0	0	0
Check	83	87	91	82	91	89

[a] Dow "2,4-Dow Weed Killer" contains 70% 2,4-D acid plus 30% $NaHCO_3$.

[b] DuPont "83% 2,4-D Weed Killer" which contains 83.5% of the sodium salt of 2,4-D (75.9% 2,4-D acid equivalent).

Fenner and Fate[1] reported abnormally large coremial growth of Ceratostomella ulmi on elm bark placed in saturated solution of 2,4-D or in the ammonium salt of this acid in concentrations of one to six grams per 800 cc. of water, and inhibition of growth of this fungus in stronger concentrations. Stevenson and Mitchell[2] reported bacteriostatic and bacteriocidal action of 2,4-D but no inhibition against Penicillium notatum (Strains 1249 and B21) and another species of Penicillium.

[1] Fenner, L. M., and L. R. Fate. Phytopath. 37: 925-928. 1947.

[2] Stevenson, E. C., and J. W. Mitchell. Science 101: 642-648. 1945.

The fumes of 2,4-D ethyl ester also exhibited inhibitory action on P. digitatum. In a limited number of tests, 1 cc. of the undiluted ester (35.2% acid equivalent) was placed on filter paper at the bottom of the moist chamber in which slides with spore suspensions were placed for germination. The spores germinated, but the germ tubes were short and distorted, and no further growth occurred. On the check slides, on the other hand, the germ tubes grew into a mass of mycelium which produced conidia by the end of 36 hours.

BOTANY DEPARTMENT, LOUISIANA STATE UNIVERSITY

BROWN BERRY DISEASE OF BLACK RASPBERRIES

J. B. Demaree

Little is known about the distribution and cause of the brown berry disease of black raspberries. It was first reported by Zundel (4) in 1941 who stated that the trouble had been prevalent in western Pennsylvania for a number of years prior to 1939 when he made some field observations. Its symptoms have been confused with those of the mild streak virus disease, yet clearly defined differences are evident.

The writer has not had an opportunity to follow the symptoms of the brown berry disease through the entire season. Apparently, the green berries become affected 2 or 3 weeks before the ripening period. The immature berry, according to Zundel, "becomes water-soaked, appearing whitish; later on when the berry is killed it turns brown." Finally, the drupelets dry out completely and the entire berry becomes hard and seedy, and adheres tightly to the receptacle. All berries in a cluster may die, turn brown and become hard, or only a few of them die and the rest develop and ripen normally. The pedicels bearing the affected berries also die, either wholly or in part.

The brown berry disease has been observed by the writer in Pennsylvania and Ohio, and it has not been reported elsewhere. Although most black raspberry plantings are free of the disease, others may have a few to many dead berries. Zundel reported heavy loss of berries in Lawrence and Butler Counties, Pennsylvania, in 1939 and 1940. He mentioned a half-acre section in one planting that on account of the prevalence of the disease yielded less than 5 quarts of fruit. He also made a careful count of the number of normal and of diseased berries in 100 berry clusters in each of 2 fields--one in Lawrence County and the other one in Butler County. In the former there were 864 berries in the 100 clusters, 81.2 percent of which were of the brown berry type. In the Butler County planting, the 100 clusters had 984 berries, of which 61.8 percent were diseased.

In 1945, Zundel published (5) a second paper in which he advanced the theory that the brown berry disease was perhaps not an entity in itself but only one expression of another but well-known black raspberry disease, mild streak, a virus disease now prevalent in some sections in New York, western Maryland (3), and western Pennsylvania, and which has been reported from Ohio (1). Jeffers and Woods (2) consider mild streak the most serious trouble affecting black raspberries in Maryland.

While circumstantial evidence during an early period in the study of the brown berry disease may have suggested a relationship of the two diseases, since both diseases affect the berries and both sometimes are

present in the same field, later observations showed that the apparent association of the two diseases in a field or on the same bush was, in the opinion of the writer, merely coincidental rather than an indication of relationship, and that brown berry and mild streak are separate and distinct diseases.

The writer has inspected black raspberry fields where the incidence of mild streak was high and brown berry symptoms were absent; and he has seen brown berry symptoms in the absence of mild streak.

In 1946 the writer accompanied Dr. Zundel and G. G. Weber, county agricultural agent, on an inspection tour of raspberry fields in York County, Pennsylvania. All of the larger plantings in the county were examined but no brown berry or mild streak was found. However, there was an outbreak of brown berry in that county the following year.

The symptoms of brown berry and of mild streak can be separated with a high degree of accurracy. The effect of brown berry is to kill one to several fruits in a berry cluster during the green stage of berry development; they then turn brown and become dry, hardy, seedy, and cling tightly to the receptacle. Unaffected berries in a cluster ripen normally. In contrast, the most distinctive symptoms of mild streak do not show until the berries are ripening, when they lack luster, are one-fifth to one-fourth smaller than normal fruits, and are poorly flavored. There is some deficiency in amount of pulp and juice; but the berries are not dry and never turn brown, as they do in the case of the brown berry disease. Also, all berries of a bush infected with mild streak show the characteristic but inconspicuous symptoms.

Zundel illustrated both diseases adequately in his 1947 revision of Extension Service Circular 250, Pennsylvania State College, entitled "Bramble Diseases." His figure 11 typifies the appearance of berries affected with mild streak and figure 12 illustrates the symptoms of the brown berry disease but the latter bears the caption "The final or 'brown berry' stage of berries affected with the mild streak in western Pennsylvania."

Literature Cited.

1. Cooley, L. M. Mild streak of black raspberries. Phytopath. 22: 905-910. 1932.
2. Jeffers, W. F. and M. W. Woods. Field studies on spread of the mild streak disease of black raspberries. Phytopath. 38: 222-226. 1948.
3. Woods, M. W. and I. C. Haut. Mild streak disease of black raspberries in Maryland. Pl. Dis. Reptr. 24: 338-340. 1940.
4. Zundel, Geo. L. A new black raspberry disease in western Pennsylvania. Pl. Dis. Reptr. 25: 87-91. 1941.
5. _____. Brown berry disease or mild streak in Pennsylvania. Pl. Dis. Reptr. 29: 567-568. 1945.

DIVISION OF FRUIT AND VEGETABLE CROPS AND DISEASES, BUREAU OF PLANT INDUSTRY, SOILS, AND AGRICULTURAL ENGINEERING, BELTSVILLE, MARYLAND

A SPOT ANTHRACNOSE OF FLOWERING DOGWOOD

Anna E. Jenkins and A. A. Bitancourt

Several specimens now available demonstrate the existence of a spot anthracnose (Elsinoë, conidial stage, Sphaceloma)[1] of flowering dogwood (Cornus florida L.), as well as its distribution in Maryland, Virginia, North Carolina, South Carolina, and Georgia. These specimens from the South, together with the circumstances under which they were taken are as follows:

Savannah, Ga., Aug. 4, 1939, A. A. Bitancourt.

This is one of the several new spot anthracnoses discovered by this collector during stops made while he was en route from Brazil to Miami, Fla., by airplane and thence by other travel to Washington, D.C. Three of these diseases have been reported previously.[2] Although the symptoms of the dogwood leaf spot connoted a spot anthracnose, neither perfect nor imperfect stage of the pathogen could be detected.

Georgetown, S. Car., Oct. 12, 1943, A. W. Blizzard.

This specimen showing spotting on a few leaves was collected during the insect and plant disease survey conducted in the general vicinity of ports of entry (1943-45). It has remained unidentified, as well as overlooked, until now.

Beltsville, Maryland, grounds of the Bureau of Plant Industry, Soils, and Agricultural Engineering, U.S. Department of Agriculture, September, 1946, A. A. Bitancourt.

Highlands Biological Laboratory grounds, Highlands, N. Car., September 2 and 5, 1947, J. A. Stevenson.

In gathering these specimens Mr. Stevenson noticed the generally diseased condition of the trees. Although the specimens were taken practically in passing, they proved of particular diagnostic value. For on the leaf spot we have found the pathogen in its perfect stage.

Athens, Ga., April 1948, J. H. Miller.

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1 Jenkins, A. E. A specific term for diseases caused by Elsinoë and Sphaceloma. Plant Dis. Rept. 31: 71. 1947.

2 Bitancourt, A. A. and A. E. Jenkins. New discoveries of Myriangiales in the Americas. Proc. Amer. Sci. Cong. 8th, Washington, 1940, 3: 149-172. 1942. illus.

Dr. Miller wrote to W. W. Diehl, Mycology and Disease Survey, as follows:

> "I am enclosing some dogwood blooms that have a blossom blight that appears to be an anthracnose. The fungus is making spores in the center of the spots. There has been a great deal of this on the streets of Atlanta and I have failed to find a description . . ."

Upon receiving this specimen Dr. Diehl made an excellent microscopic mount, which he asked one of us (Jenkins) to view. The hyaline hyphomycetous development is consistent for Sphaceloma[3], as is the bract spot for a spot anthracnose. Dr. Miller has written (May 14) that the trees have passed their blossoming at Athens, but that the leaves now show the spotting.

Norfolk, Va., May 9, 1948, H. T. Cook.

Dr. Cook, upon being advised personally of the existence of a spot anthracnose of flowering dogwood and its symptoms, immediately remarked that he was familiar with such a disease; not only had he observed it on blooms and leaves of a tree in his own grounds, but specimens had been sent for diagnosis to the Virginia Truck Experiment Station. The spots were sterile, and the disease remained undiagnosed. The specimen cited above shows typical leaf spots and also cankers on the peduncles.

The symptoms of the disease as here delineated are based chiefly on dry specimens (fig. 1).

On leaves, spots are scanty to abundant up to 100 or more on a single blade. They are scattered over the surface including the leaf margin or may be disposed on or along the midrib. Individual spots are circular to angular or irregular, reaching 1 mm., less often 1.5-2 mm. in diam. Above, where they are more prominent and may be slightly raised at the margin, they are "reddish gray"[4] becoming "weak yellowish gray,"[4] at the center. This thin often translucent pale tissue readily fragments away, resulting in shot hole effects or skeletonized areas. Parts of the leaf, such as the apex, all at one side of the midrib, or all but a spatula-shaped central part with the midrib as the axis, also may be completely severed.

On petioles, peduncles, and small stems, cankers correspond more or less closely to the blade spot. They are circular to elliptical, flat or somewhat elevated or slightly depressed.

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[3] Jenkins, A. E. and A. A. Bitancourt. Revised descriptions of the genera Elsinoë and Sphaceloma. Mycologia 33: 338-340. 1941.

[4] Color according to ISCC-NBS system of color designation (Judd, D. B. Method of designating colors. [U.S.] Natl. Bur. Standards Jour. Res. (RP 1239) 23:355-385. 1939. illus. For these color determinations we are indebted to Josephine J. Tomaszewski, Cotton Branch, Research and Testing Division, PMA, U. S. Department of Agriculture.

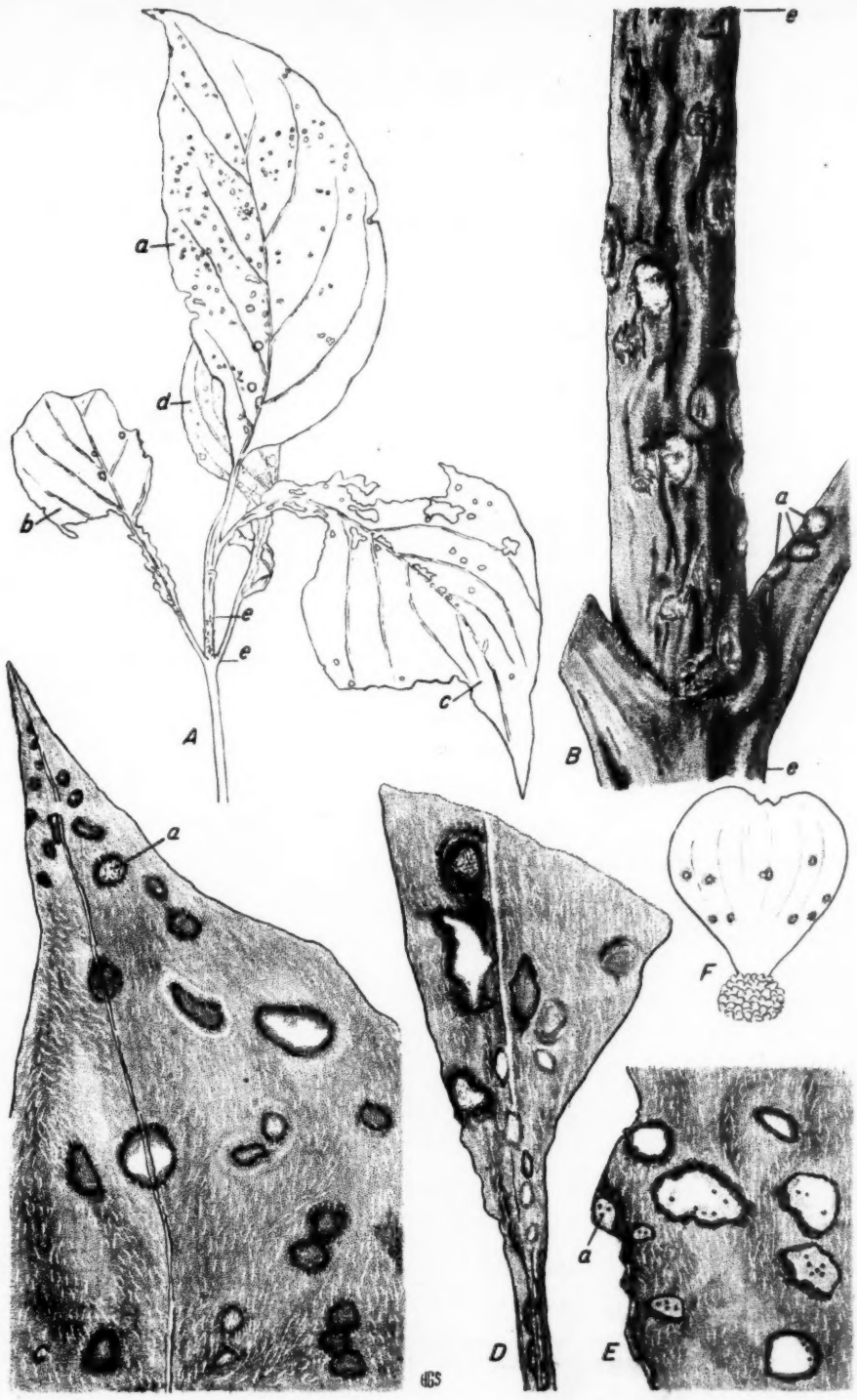

Fig. 1. Spot anthracnose of flowering dogwood. A, Shoot with severely diseased leaves (*a* - *d*) and heavily cankered stem area (*e e*); *d*, lower leaf surface; X 1. B, *a*, petiole lesions; *e e*, cankers in A, *e e*; X 13. C - E, Various leaf spots on upper third of a young leaf (C) and on mature leaves (D and E); D, basal part including grooved petiole; E, intermediate area including leaf margin; C, *a*, developing ascomata (small dots) of the *Elsinoë*; E, *a*, mature ascomata; X 13. Drawings by H. G. Stueler.

On the floral bracts, the spots are few to numerous, scattered, spherical to elliptical, reaching about 1 mm. in diameter. Often they occur singly, but they may be present in small groups and may appear on or near the veins. Of more delicate coloration than the other lesions, they are light tan in the central area, and, characteristically, are delimited by a narrow brownish to purplish brown border, outside which is an indistinct tan zone.

On some of the leaf spots are one to several dark punctiform areas, --ascomata, of the pathogen (Elsinoë).

It is hoped that this account will aid in diagnosing this heretofore obscure disease of one of the most beautiful of North American flowering trees--in this case the official state tree and flower of Virginia.

The writers will appreciate receiving specimens that appear to be the spot anthracnose. Such collections should be pressed dry before sending.

DIVISION OF MYCOLOGY AND DISEASE SURVEY, BUREAU OF PLANT INDUSTRY, SOILS, AND AGRICULTURAL ENGINEERING, BELTSVILLE, MARYLAND; AND INSTITUTO BIOLOGICO, SÃO PAULO, BRAZIL.

TWO NEW NEMATODE DISEASES OF THE AFRICAN VIOLET[1]

A. C. Tarjan and C. E. Cox

From a group of four African Violets, Saintpaulia sp., showing stunting and foliar wilt symptoms, two plants were found infected with the Meadow Nematode, Pratylenchus sp., while one was found infected with the Root-Knot Nematode, Heterodera marioni. In so far as the authors have been able to determine, this is the first time either of these parasites has been reported on Saintpaulia sp.

The Meadow Nematode was found on two plants of the "ionantha" variety. The basal foliage of these plants was wilted with the long, flaccid petioles of the leaves hanging over the edge of the pots. The younger leaves were more upright in growth habit than normal, and the leaf blades had failed to expand normally giving the plants a dwarfed appearance (Fig. 4). Inspection of the underground parts revealed a stunted root system showing chracteristic Meadow Nematode injury consisting of brownish discolored roots, the distal parts of which had decayed and sloughed off leaving strands of the vascular cylinder exposed.

The Root-Knot Nematode was found on a plant of the "Plum" variety. Above ground symptoms consisted of a compact, dwarfed habit of growth, pronounced downward curling or rolling of the margins of the leaves, and wilting of the basal leaves. The petioles and foliage were less flaccid, however, than in the case of the Meadow Nematode infected plants (Figs. 1-2). Inspection of the roots revealed a dwarfed condition with brownish discoloration (Fig. 3). The galls were small and generally fusiform in shape. There was marked proliferation of lateral roots especially in the vicinity of the galls. In some cases these short lateral roots arose from the galls themselves and in other cases from above and below the swellings.

Having seen so few plants infected by these two nematodes, it is impossible to state whether the symptoms described represent the entire symptom range or whether infection with the absence of certain of these symptoms is possible.

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1 Miscellaneous Publication No. 72. Contribution No. 2115 of the Maryland Agricultural Experiment Station (Department of Plant Pathology).

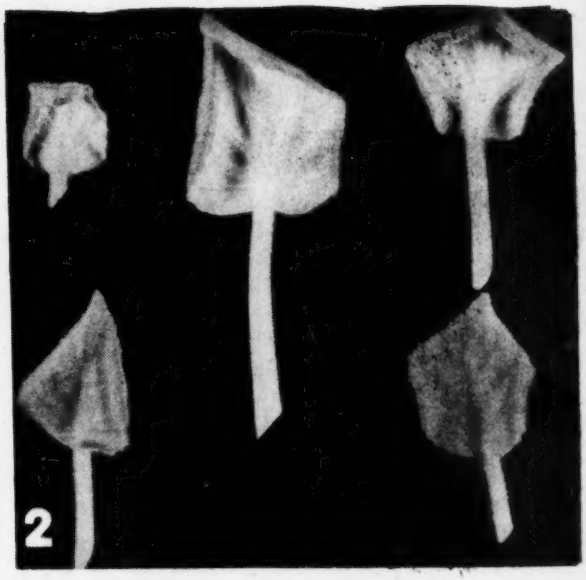

Fig. 1 African Violet infected by the Root-Knot Nematode.

Fig. 2 Leaves from the plant in Fig. 1 showing the characteristic downward curling of the leaves.

Fig. 3 Plant shown in Fig. 1 showing dwarfed condition of the root system.

Fig. 4 African Violet infected by the Meadow Nematode.

TREATMENTS FOR GLADIOLUS CORM ROTS IN WASHINGTON: PROGRESS REPORT FOR 1947

Charles J. Gould

Since a lack of time prevents preparation of a complete report of the 1947 gladiolus corm treatments, the following summary is being released for possible use by others preparing experimental programs in 1948. None of the statements made herein should be interpreted by growers as recommendations, for it is becoming increasingly evident that materials effective and practicable in one area for a certain disease on gladiolus will not necessarily be effective and practicable in another area against the same diseases, and particularly against different diseases. As an example, the most effective materials in western Washington are not the most effective in eastern Washington. These areas are, of course, geographically close, but climatically quite distinct. The diseases are also, to some extent, different.

Fourteen materials were tested in 1947, usually at more than one concentration, and in some cases on more than one variety. Most treatments were applied as 15-minute dips just prior to planting. Five replications of 100 to 200 number 5 corms were used in most of the tests.

The following materials and concentrations were at least partially effective for controlling corm rots (especially Dry Rot) in western Washington: Calogreen (1 lb/8 gal; 1 lb/10 gal); Tersan (1 lb/6 gal; 1 lb/9 gal); Arasan SF (1 lb/9 gal); Dowicide 9-B (1 lb/21 gal); Puratized Agricultural Spray (1:10,000 active); Puraturf 177 (1 lb/12 gal); New Improved Ceresan (1 lb/50 gal); Ceresan M (1 lb/60 gal); No. 307[1] (1 lb/12 gal); and Lysol (1 qt/50 gal).

The three materials that gave best control of Fusarium and Dry Rots in eastern Washington were: New Improved Ceresan (1 lb/50 gal); Ceresan M (1 lb/60 gal), and Supergermite (1 qt/50 gal). Tersan and Calogreen were also tested but were not so effective.

The following additional facts were also obtained in the 1947 tests.

1. Mercury compounds, in general, definitely delayed flowering in western Washington. On the other hand Tersan and Arasan SF usually accelerated flowering. Such an effect upon flowering is of considerable importance to western Washington growers in view of their sales of cut flowers.
2. Calogreen, in general, effectively controlled Dry Rot. However, in

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[1] 2, 4, 5-Trichlorophenyl acetate, obtained from Dr. J. H. Standen, Boyce Thompson Institute, Yonkers, New York

tests with two Fusarium-infected stocks, the Fusarium brown rot was greater in Calogreen-treated sets than in the untreated sets in both eastern and western Washington. Plants of the Greta Garbo variety grown from corms treated with Calogreen had narrower leaves than those in untreated sets.

3. Tersan at 1 lb/6 gallons was somewhat more effective than Arasan SF at 1 lb/9 gallons.

4. Puraturf 177 (at 1 lb/12 gal) was one of the most promising of the newer materials tested, giving results in flower cut, corm yield and freedom from rot, comparable to Tersan (at 1 lb/6 gal).

5. Corms treated in December with Dowicide 9B or Tersan developed slightly less rot than similar sets treated at planting time in April.

6. None of the fungicidal treatments was effective in controlling Botrytis corm rot. The best control of this rot seems to be the artificial drying of corms immediately after digging.

7. Results of numerous cormel-treatment tests were inconsistent. In certain tests, untreated sets that were soaked in water before planting, gave greater yields than the treated sets.

As a result of the tests in 1947 and preceding years, the following suggestions and comments are being released to Washington gladiolus growers:

Gladiolus Corms

1. Discard all diseased corms.
2. Dip corms, before planting, as follows:

In Western Washington for Dry Rot

A. For lightly-infected stocks use Tersan (1 lb/6 gal. for 5-15 minutes).
B. For severely-infected stocks use Calogreen (1 lb/8 gal. for 5-15 minutes).
(Calogreen delayed flowering in certain varieties last year and did not control Fusarium rot, but it was generally very effective against Dry Rot.)
C. For stocks "imported" from other areas that might contain Fusarium rot, use one of the materials recommended for Eastern Washington.

In Eastern Washington
for Dry Rot and Fusarium Rot

D. Lysol or Supergermite (1 qt/50 gal. for 3-6 hours), or
E. New Improved Ceresan (1 lb/50 gal. for 15 minutes).
F. The new compound Ceresan M (1 lb/60 gal. for 15 minutes) also gave promising results last year.

Gladiolus Cormels

1. Save cormels from lightly-infected or clean stocks only.
2. Although the treating tests run last year were on a fairly large scale, the results were inconsistent. In many cases the samples of cormels that were soaked only in water before planting gave the best results. If a fungicidal treatment is desired, try one of those listed above for corms. Varieties may respond differently to different treatments.

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THE HISTORY, DISTRIBUTION, AND NOMENCLATURE OF THE ROSE BLACK-SPOT FUNGUS

Kenneth F. Baker[1]

The early history of black spot, the most important disease of rose, has not been presented although more than 450 papers on the pathogen have appeared. For example, the first American paper commonly cited is that of Scribner (97) in 1888, although Farlow's bibliography (39) lists numerous earlier reports for North America. Only first records or otherwise interesting reports from various areas are presented in order to limit an already lengthy bibliography.

Records in Europe

The first definite report of the fungus was that of Fries in his Observationes Mycologicae in 1815 (42). This collection on rose leaves in Sweden by Erik Acharius apparently was called Sclerotium radiosum Achar. Fries named it Erysiphe radiosum Fr. and stated that the leaf spots were thin, becoming fuscous, 1/4-5/12 inch in diameter, with dusky, slow-growing, stellate, radiating, fibrous strands, and with small black heads which were not set free.

Libert (65) in 1827 gave the first description and illustrations that unquestionably refer to the blackspot fungus. She called it Asteroma rosae Lib., and listed Himantia pulchella as a possible synonym. It occurred principally on Rosa francofurtana Muench. (R. turbinata)[2] in fall near Malmédy, Belgium. The fungus was reported near Louvain in 1835 by Kichx (60).

In the Elenchus Fungorum of 1828 (43) Fries expanded his earlier description, but placed the binomial, E. radiosa, in quotation marks. The fungus was stated to be common in leaves of Rosa centifolia L. and R. gallica L. in the fall. In the Systema mycologicum of 1829 (44) he mentioned "E. radiosum" and cited his Observationes and Elenchus, as well as the paper of Libert. He did not, however, use the binomial Asteroma radiosum Fr., as has been said (e.g. 21, 48). In his Summa vegetabilium Scandinaviae of 1849 (45) he placed the fungus in a new genus of the Pyrenomycetes as Actinonema Rosae (Lib.) Fr., and this name has been the one most commonly used down to the present day. The genus Asteroma has come into poor standing (51), and the name Asteroma rosae generally abandoned.

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1 It is a pleasure to acknowledge the use of the excellent library facilities of the Department of Plant Pathology, Cornell University, in this study.

2 The nomenclature follows L. H. Bailey's "The Standard Cyclopedia of Horticulure." Macmillan, New York City. 1922.

The disease was known in France by 1822, for Persoon (84) listed Himantia pulchella Pers. on Rosa gallica in the country around Paris. Link (67) apparently expanded this description and applied the binomial, Sporotrichum macularum Link in 1824. Duby (34) in 1830 reported S. pulchellum Duby in the same area, and cited H. pulchella Pers. and S. macularum Link as synonyms. Dothidea Rosae Duby was also described on Rosa francofurtana (R. turbinata), with Asteroma Rosae Lib., Himantia pulchella Pers., and Sporotrichum macularum Link as synonyms. These binomials of the fungus were all regarded as synonymous by Wallroth (112). Grognot (50) reported Asteroma rosae Lib. and A. penicillatum Grog. in France in 1863. The latter he stated was the same as A. rosae forma rosae semperflorens attributed to Desmazières, and was placed under A. rosae presumably as a synonym. A specimen of this (Roumeguère, Fung. Sel. Galliaei Exsic. No. 239) is definitely the black-spot fungus.

In Germany what seems to have been the black-spot fungus was reported by Wallroth (112) in 1833 as Phlyctidium Rosae Wallr. on Rosa gallica (R. chamaerrhodi var. gallica) in the fall. He listed Asteroma stellatum Wallr., Dendrina Rosae Fr., Dothidea Alphitomorpha Wallr., Asteroma Rosae Lib., and Erysiphe radiosum Fr. as synonyms, inter allia; the first two binomials have not been found elsewhere. Bonorden (12) described the fungus as Dicoccum Rosae Bon. in Germany in 1853. His description and illustrations clearly refer to the black-spot fungus and were so recognized by Saccardo (93, vol. 10: 477) and by Briosi and Cavara (16) who revised the binomial to Marsonia rosae (Bon.) Briosi et Cav. in 1889, and identified it with Actinonema rosae Fr. (41). Döll (32) listed Sporotrichum pulchellum Duby in a German rose manual in 1855. Fuckel (47) reported the disease in 1860, and again (48) in 1869 as common on R. centifolia and infrequent on R. canina L. in the fall. The latter may be the first report of the fungus on a wild rose in its natural habitat (41).

Berkeley (7) reported Asteroma Rosae Lib. as very common in the autumns of 1840 and 1841 in England, and again (8) in 1860. In Scotland it was reported by Stevenson (105) in 1879. Trail (110) collected it in September, 1888 on R. setigera var. tomentosa Gray (R. tomentosa) in Scotland and named it Marsonia Rosae Trail, without referring to any earlier description. Saccardo (93, vol. 10: 477) recognized this as Actinonema rosae.

The fungus was reported (33) as Sporotrichum macularum Link in Holland in 1844. Rostrup (66) apparently first collected the fungus in Denmark in July 1866. It was found by von Thümen (109) near Athens, Greece in 1871. The earliest report found for Italy is that of Saccardo (92) in 1880; others were in 1884 (93, vol. 3: 408) and 1889 (16, 21). It was observed in Hungary (38), and in Austria and Portugal (93, vol. 3: 408) by 1884. In Switzerland it was collected at Bern by von Tavel in 1894, and reported again in 1902 (41). The disease was reported in south-

central Russia in 1907 (86), in Czechoslovakia in 1925-26 (99), in Bulgaria in 1930 (5), and in Poland in 1931 (63). Borg (13) stated that the pathogen was introduced to Malta about 1932-33. In Rumania it was observed in Bucharest in 1930 (94, and Savulescu, Herb. Mycol. Romanicum Fasc. VI, No. 272), in Jugoslavia by 1937 (18), in Turkey by 1947 (14).

Records in North America

The early records of the black-spot fungus in this country were contemporaneous with those of Europe. In a report to the American Philosophical Society in Philadelphia on April 15, 1831 Schweinitz (95) described it as "Dothidea Rosae L.v.S. in senescent leaves of cultivated rose in a Philadelphia garden. Spots round, determinate, with definite margins, 1/4 inch in diameter, often coalescent, epiphyllous, fuligenous to dusky black; context of thick, branched, radiating strands; central cavity large, flattened, moist, somewhat cup-like, at first with white contents." He likewise placed the fungus in the Pyrenomycetes. Saccardo (93, vol. 2: 611) suggested that this might be an Actinonema. Schweinitz' specimen No. 1941 in Philadelphia was examined by Ellis (37) and identified as Actinonema Rosae Lib.; the specimen (No. 9276) in the Berkeley Herbarium was identified by Cooke (23) as Asteroma Rosae Fr. There can be no doubt that black spot was present at least by the spring of 1831. Because it is unlikely that the disease would be evident before mid-April in Philadelphia, the specimens probably were collected in the fall or summer of 1830.

According to Farlow (39) a specimen (Fungi Caroliniana Exsiccati, Fasc. 1, No. 68) distributed by H. W. Ravenel in 1852 as Dothidea Rosae Schwein. is the black-spot organism. Cooke (22) examined a later Ravenel specimen (No. 2115) collected at Aiken, South Carolina on rose, and named it Asteroma Rosae Fr., but gave no date. Berkeley (9) reported in 1874 a specimen from Ravenel (No. 3676) of Asteroma Rosae Lib. and an "A. punctiforme Berk." on rose leaves from South Carolina. A well-preserved specimen (Fungi Americana Exsiccati No. 520; Asteroma Rosae DC. on rose leaves, Aiken, S. Car.) in the Mycological Herbarium, Cornell University, definitely is the black-spot fungus. Apparently this group of exsiccati was distributed in 1881 by Cooke as Asteroma Rosae Lib., but specimens must have been sent to England by 1874. Parsons (81) in an American rose manual in 1856 listed Sporotrichum pulchellum Duby as a parasite. Curtis (26) in 1867 reported Asteroma Rosae Lib. and an "A. geographica Fr." on rose leaves in North Carolina. Frost (46) reported Dothidea Rosae Fr. as an Ascomycete on rose in New England in 1868. Peck (83) reported a collection of Asteroma Rosae DC. on leaves of R. rubiginosa L. collected in New York by G. W. Clinton in October. Since the paper was dated January 13, 1875, this collection clearly was October, 1874 or earlier.

Other early reports in the east and south may be mentioned. Scribner

(96) stated that the disease was damaging to outdoor roses in the District of Columbia in 1886. Maynard (70) reported the disease on greenhouse roses in Massachusetts in December 1887. It was reported (64) in southern Louisiana in 1887, and at Ocean Springs, Mississippi in 1889 (35). A specimen (No. 6750) of the fungus in the Mycological Herbarium, Cornell University, was collected in New Jersey in August 1880 by G. F. Meschutt, and it was recorded (36) in two places by 1889. In Alabama it was found on cultivated roses in 1890 (6). It was reported in Connecticut (107) and Delaware (Ellis and Everhart, Fungi Columb. No. 277) in 1893, and in Vermont the disease was collected in 1894 by L. R. Jones (77). In West Virginia, Nuttall (73) collected it in 1894. In 1900 the fungus was reported in Maine (89) and Ohio (98).

In the Central States the disease was observed in Indiana (111) by Barnes in January 1887; this record at Tippecanoe may be the first greenhouse report in this country. In Nebraska the fungus was said (88) to be common on cultivated roses in 1889, and in Texas (58) in 1890. The fungus was collected in Missouri in 1893 (Seymour and Earl, Economic Fungi No. 469) and reported before 1902 (82), in Kansas in 1906 (Bartholomew, Cornell Mycol. Herb. No. 18610), and in Wisconsin by 1912 (27).

In the western part of this country apparently the earliest report is that of Harkness and Vize (No. 723) in California sometime prior to 1878 (53) as Asteroma rosae DC. Harkness and Moore (54) listed Dothidea Rosae Fr. on rose in their Catalogue of 1880 in Sac[ramento?]. and A. Rosae DC. in San Francisco. The fungus was collected (71) on R. californica Cham. & Schlecht. near Pasadena in southern California in 1893-5. Actinonema rosae Lév. (sic) was reported (59) on R. blanda Ait. or R. acicularis var. bourgeauiana Crepin (R. sayi) at Helena, Montana by 1889.

The disease has been reported in all states in this country except Arizona, Wyoming, and Nevada (69, 106); a letter of March 30, 1948 from Dr. P. R. Miller stated that the Plant Disease Survey records on this were still unchanged.

In Canada, the disease was common in Ontario at least by 1911 (57), in Manitoba by 1929 (10), and in Saskatchewan by 1938 (11); it has since been reported (25) in British Columbia, Quebec, New Brunswick, Nova Scotia, and Prince Edward Island.

Records in Miscellaneous Areas

The fungus was collected in South China at a fairly early date (October 1910) on R. laevigata Michx. (74). It was collected again in 1927 (108) and in 1934 (79, and Rel. Herb. Fung. Sinensis No. 2163). Stevens and Roldan (104) reported that "the disease was unknown in the

Philippines until November 22, 1930, when a specimen was sent from Cavite." Cooke (24) reported the fungus in Victoria and Queensland, Australia in 1892. It was recorded in South Australia in 1912 (78) and in New South Wales in 1939 (2). The disease was recorded in Ceylon by 1928 (1). In Hawaii, the fungus was reported in 1936 (80), but had not been listed by Stevens (103) in 1925. The pathogen was present in the United Provinces, India by 1941 (40), and in New Zealand the fungus was first reported in the period 1942-46 (15).

The pathogen apparently was first reported in South America in Argentina in 1880 (100), in Paraguay in 1881 (101), and in São Paulo, Brasil in 1888 (102) by Spegazzini. It was recorded from northeastern Brasil in 1936 (85). A specimen (Fungi Porto Rico No. 664) of the fungus was collected in Puerto Rico in March 1916 by H. H. Whetzel and E. W. Olive. It was present in Trinidad (90) in 1919 and British Guiana (3) in 1926. Chardon and Toro reported the disease in Colombia in 1929 (19- and Venezuela in 1930 (20). It was collected in Bermuda (Whetzel, Bermuda Fungi No. 157) in April 1922, and was said to be common by 1935 (91). The disease was observed in the Dominican Republic in 1936 (49).

In Africa the disease was reported in Egypt in 1920-22 (17), South Africa in 1924 (31), Kenya in 1936 (72), Uganda in 1937 (52), and Rhodesia in 1938 (56).

Names of the Pathogen

The following names have been applied to the acervular or imperfect stage of this fungus in the literature:

Sclerotium radiosum Acharius	1815	(42)
Erysiphe radiosum Fries	1815	(42)
Himantia pulchella Persoon	1822	(84)
Sporotrichum macularum Link	1824	(67)
Asteroma rosae Libert	1827	(65)
Dothidea Alphitomorpha Wallroth	1828	(43, 112)
Dothidea Rosae Duby	1830	(34)
Sporotrichum pulchellum Duby	1830	(34)
Dothidea Rosae Schweinitz	1832	(95)
Phlyctidium Rosae Wallroth	1833	(112)
Asteroma stellatum Wallroth	1833	(112)
Dendrina Rosae Fries	1833	(112)
Actinonema rosae (Lib.) Fries	1849	(45)
Dicoccum Rosae Bonorden	1853	(12)
Asteroma rosae DeCandolle (*sic*)	1860	(8, *et al.*)
Asteroma penicillatum Grognot	1863	(50)
Dothidea rosae Fries (*sic*)	1868	(46, *et al.*)
Asteroma radiosum Fries (*sic*)	1869	(48, *et al.*)

Phyllachora rosae (Schw.) Saccardo	1883	(93, vol. 2: 611, et al.)
Marsonia rosae Trail	1889	(110)
Marsonia rosae (Bon.) Briosi and Cavara	1889	(16)
Phyllachora impressa Saccardo	1891	(21)
Marssonina rosae Trail	1912	(28)
Marssonina rosae (Lib.) Lind	1913	(66, et al.)
Marssonina rosae (Lib.) Diedicke	1915	(29, et. al.)

Establishment of the true synonymy of some of the older binomials would require examination of specimens, if they are still existent.

The correct name of the imperfect stage seems to be Marssonina rosae (Lib.) Lind; it is in accordance with the International Rules of Botanical Nomenclature, Section 9, Article 54. It is often used, particularly by European mycologists (e.g., 28, 29, 41, 63, 66, 76, and the Commonwealth Mycological Institute, Kew), although the authorities cited may be different.

Acharius' binomial is a nomen nudum. Erysiphe radiosum Fries was so briefly described as to leave its identity in doubt, and was rejected by Fries in favor of Libert's species in creating Actinonema rosae (Lib.) Fries. About the identity of Himantia pulchella Pers. there is doubt, and the same is true of Link's Sporotrichum macularum based on it. Thus, Libert's Asteroma rosae seems to be the earliest valid name for this fungus.

Fries interpreted the fruiting body as a closed structure and erected the genus Actinonema in his Pyrenomycetes. Although the summer conidia quite generally (e.g., 30, 41, 114) are considered to be borne in acervuli, the original pycnidial name usually has been used. Some have preferred to call it Marssonia for that reason. Because Marsonia (and its variant, Marssonia) is a later homonym of a genus of the Gesneriaceae, Magnus (68) transferred species of Marssonia to a new genus, Marssonina. The change has been generally accepted, and Marssonina listed in the nomina generica conservanda of the 1935 International Rules. The rose fungus was not transferred by Magnus because it was then commonly placed under Actinonema. Von Höhnel (55) apparently first suggested the transfer of Actinonema rosae to Marssonina in 1907 but did not formally make the new combination. Potebnia (87) made a similar but more indefinite suggestion.

Diedicke (28) made a formal but erroneous transfer in 1912, citing it as Marssonina rosae (Lib.) Trail, presumably because he interpreted Trail's binomial as a transfer to Marsonia and therefore to Marssonina. There is no evidence that Trail was aware of other names, and Diedicke (29) recognized this in 1915 by citing himself as authority. The first valid transfer, therefore, seems to be that of Lind (66) in 1913[3].

[3]The kind counsel of Dr. G. R. Bisby (correspondence of March 30, 1948) has been followed in this matter.

Grove (51) corrected the original misconception by transfer of Actinonema Fries from the Sphaeropsidales to the Melanconiales, and by "emendation" and retention in it only of A. rosae and A. aquilegiae, forms with a "definite fibrillose subiculum". All other Actinonemas, including A. fragariae, were placed in Marssonina. Such an action would be justified only if there was valid basis for separation of Marssonina and Actinonema. However, it has been observed (41) that (a) the rose fungus does not always have macroscopically visible subcuticular fibrillar mycelium (e.g., in infections of leaf undersurfaces, petioles, or young shoots), and (b) these fibrillose structures occur also in species (e.g., M. fragariae, M. populi) placed by Grove in Marssonina. There seems, therefore, no valid reason for reviving the genus Actinonema, from which all species had already been transferred to Marssonina. Nannfeldt (75) favored discarding the genus Actinonema.

The situation of the winter conidial structures is interesting. These were described (30) as having no side walls and being "subepidermal acervuli", but with the statement that "one might prefer to call them pycnidia." Wolf and Wolf (114) state that subepidermal apothecia sometimes bear conidia as well as asci, and Aronescu (4) referred to them as "apothecial conidia". Frick (41) considered that the structures might be classed under Excipulaceae of the Sphaeropsidales, but that no special name was required. It would seem that the difference between summer and winter asexual structures is a result of parasitic versus saprophytic growth and of the relative depth of host penetration attained. Similar structures have been reported for Diplocarpon earliana (62), D. agrostemmatis (76), D. Soraueri (61, 76), and Drepanopeziza ribis (61), and there is a tendency to their development in the Drepanopezizoideae of Nannfeldt (76). Nannfeldt (75) stated that location of the structures in the host, and the presence of fibrillose hyphal strands were not good generic characters. "Thus the name Marssonina should be used for all subcuticular, intra- or subepidermal Melanconiales with hyaline, uniseptate conidia, which are never fusiform or cylindrical, and genetically connected with Drepanopeziza or allies" (75). The fungus does not form pycnidia in culture (41), but when old tends to form slimy conidial masses which sometimes resemble acervuli. Acervuli are produced on bean pods (113).

The correct name of the perfect stage is Diplocarpon rosae Wolf (see footnote 3), and is usually so cited. This stage was described in 1912 (113) under this binomial, but sometimes (e.g., 38, 41, 108) it is cited with Libert or Fries as first authority. Under the International Rules, Section 10, Article 57, the first valid name applied to the perfect state is binding. Under the type method (Section 2, Article 18), the binomial of the perfect state applies only to the type specimen of that stage, and is not considered as a transfer of the name of the imperfect state even if the same specific epithet is used. Apparently the perfect stage has been encountered only in the northeastern United States (4, 113) and south-central Canada (11).

Discussion and Summary

The fungus which causes black spot of rose was first recorded in Sweden in 1815, but soon was reported in France (1822), Belgium (1827), United States (1830), Germany (1833), England (1840), and Holland (1844). It has subsequently been reported in Denmark (1866), Greece (1871), Scotland (1879), Italy and Argentina (1880), Paraguay (1881), Hungary, Portugal, and Austria (1884), São Paulo, Brasil (1888), Victoria and Queensland (1892), Switzerland (1894), south-central Russia (1907), China (1910), Canada (1911), South Australia (1912), Puerto Rico (1916), Trinidad (1919), Egypt (1920-22), Bermuda (1922), South Africa (1924), Czechoslovakia (1925-26), British Guiana (1926), Ceylon (1928), Colombia (1929), Venezuela, Bulgaria, Rumania, and Philippine Islands (1930), Poland (1931), Malta (1932-33), Hawaii, Dominican Republic, northeastern Brasil, and Kenya Colony (1936), Jugoslavia and Uganda (1937), Rhodesia (1938), New South Wales (1939), India (1941), New Zealand (1942-46), Turkey (1947). The fungus is now distributed through the temperate and tropical regions of the world. The earliest collectors in various parts of the United States found the pathogen already established. It was present in the Northeastern States by 1830, Southeastern States by 1852, Central States by 1887, Mountain States by 1889, Southwest by 1878, and Northwest by 1889.

In Europe, the early mycologists (e.g., Schaeffer [1762-1774; 1800], Bolton [1788], Tode [1790-91], Bulliard [1791], and Sowerby [1797-1803]) did not report the black-spot fungus, but they generally studied the larger and more conspicuous forms.

The roses grown before 1850 were either wild species or not far removed from them. The Hybrid Perpetuals were introduced in 1843 and dominated gardens until 1890. The Hybrid Teas were introduced in 1867 and became prominent after 1890. Pernetianas appeared in 1900. These types were more susceptible to black spot than those used earlier, and it is natural that the disease should then receive increasing attention.

Because of the already wide distribution of the fungus in the first records, it is probable that it was disseminated at a much earlier date. Only four instances were found in which the fungus seems to have definitely been introduced in historic times -- Philippine Islands, Malta, Hawaii, and New Zealand.

There is nothing about the symptoms produced that is sufficiently distinctive to permit positive identification from ancient herbals, as was the case with tulip mosaic. The rose has been cultivated since the earliest times, and wild species occur over much of temperate Europe, Asia, and North America. Because of these facts it is improbable that the native home of the fungus can now definitely be located. It has been suggested (41) that the fungus probably originated in North America, because that is where the perfect stage occurs. However, it

seems just as reasonable to suggest that it originated in Europe or Asia because that is where the only well-tested immune species, Rosa cinnamomea L. and R. pendulina L., thus far reported (41), and the highly resistant ones, R. arvensis Huds. (41), R. multiflora Thunb., and R. wichuraiana Crepin, are native.

As would be expected with a fungus of such early general distribution and conspicuous destructiveness, the pathogen has been given many names: 25 were found that have been applied to the conidial stage.

The correct binomial for the imperfect stage is Marssonina rosae (Lib.) Lind, and that for the perfect stage is Diplocarpon rosae Wolf. The perfect stage has been reported only in the United States and Canada.

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BRIEF NOTES ON PLANT DISEASES

BACTERIAL CANKER OF COWPEA IN ILLINOIS

By G. H. Boewe

A bacterial disease of cowpea new to the State was collected on September 24, 1947, in Calhoun County in southwest-central Illinois. This disease appears to be identical with bacterial canker of cowpeas reported by Dunlap (PDR 27 (14): 274. 1943), Hoffmaster (Phytopath. 34 (4): 439-441. 1944), and Lefebvre and Sherwin (Phytopath. 35 (6): 487. 1945), and described as a new species by Burkholder (Phytopath. 34 (4): 430-432. 1944). The seed used in this six-acre field was a mixture, consisting chiefly of New Era, Red Rippers, and Whippoorwill. Approximately 0.5 percent of the plants were diseased. It was impossible to tell what variety was affected as no pods were present on the diseased plants.

ILLINOIS NATURAL HISTORY SURVEY, URBANA

TWO PLANT DISEASES NEWLY REPORTED IN GEORGIA

By Julian H. Miller

Azalea flower-spot (Ovulinia azaleae) has been found in a garden at Columbus, Georgia. This is the first specimen I have received in this State. Both red and white forms were heavily infected in March. No conidia were found on the petal spots when the specimens arrived, but sclerotia formed abundantly in petri dishes.

A leaf spot of wheat (Helminthosporium tritici-vulgaris) has also been found for the first time by the writer in this State. A field of Sanford wheat in Oconee County was heavily infected with small circular leaf spots. The entire field had a yellow appearance. This observation was made about the middle of March, and at the present time, one month later, the plants appear to have outgrown the disease.

UNIVERSITY OF GEORGIA, ATHENS

CHECK LIST REVISION

Freeman Weiss

HELICHRYSUM (COMPOSITAE)

HELICHRYSUM BRACTEATUM Andr., STRAWFLOWER. Annual herb of Australia, widely grown as an everlasting flower.

Fusarium sp., stem rot. Wash.
Heterodera marioni (Cornu) Goodey, root knot. ?Fla.
Verticillium albo-atrum Reinke & Berth., wilt. Calif.

Curly top -- virus (Chlorogenus eutetticola Holmes, Beta virus 1 K. M. Sm.). Calif., Ore., Wash.
Yellows, bunchy top -- virus (Chlorogenus callistephi Holmes, Callistephus virus 1 K.M.Sm.). Eastern and Central States. Also var. californicus Holmes, Callistephus virus 1A K.M.Sm., in Calif.

HELIOPSIS (COMPOSITAE)

HELIOPSIS HELIANTHOIDES (L.) Sweet (1), and H. SCABRA Dunal (2), OX-EYE. Perennial herbs of the Eastern and Central States and southward, sometimes grown in gardens and useful to wildlife.

Coleosporium helianthi (Schw.) Arth. (II,III), rust. N. Car. (2). O and I on Pinus spp.
Erysiphe cichoracearum DC., powdery mildew. Iowa, Minn., Nebr., N.Mex., N.Dak., Wis. (2).
Gibberidea heliopsidis (Schw.) Shear, black patch on leaves and stems.
Phyllosticta pitcheriana Fairm., leaf spot. N.Y. (1)
Phymatotrichum omnivorum (Shear) Dug., root rot. Tex.
Puccinia batesiana Arth. (0,I,III), rust. Del., Pa. (1); Iowa, Kans., Md., Minn., Nebr., Wis. (2).
P. helianthi Schw. (0,I,II,III), Ind., Minn., N.Car., Pa., Va. (1); Ind., Iowa, Minn. (2)
Septoria helianthi Ell. & Kell., leaf spot. Ind. (1)
S. heliopsidis Ell. & Dearn. Wis. (2)

Mosaic -- unidentified virus. Iowa.

HIERACIUM (COMPOSITAE)

HIERACIUM spp., HAWKWEED, especially H. AURANTIACUM L., ORANGE H. (1), H. CANADENSE Michx. (2), H. VENOSUM L. (3), other spp. (4).

HIERACIUM cont.

Numerous perennial herbs native in the U. S. or naturalized from Europe; some occasionally grown in rock gardens, several are useful to wildlife; (1) is a troublesome weed in meadows in the Northeast.

Aecidium columbiense Ell. & Ev. (0,I), rust. N.Y. (3); Calif., Ida., Ore., Wash. (4). II and III unknown.
Bremia lactucae Regel, downy mildew. Wis. (1)
Cercospora hieracii Ell. & Ev., leaf spot. Ala., N.Car. (3)
Erysiphe cichoraceaarum DC., powdery mildew. Tex. (1), Ill. (2), Pa., Wash. (4)
Phoma hieracii Rostr., stem blight. Tex. (1)
Phyllosticta decidua Ell. & Kell., leaf spot. Wis. (1)
Phymatotrichum omnivorum (Shear) Dug., root rot. (Tex. 1,4)
Puccinia extensicola Plowr. var. hieraciata (Schw.) Arth. (0,I), rust. Ill., Wis. (2); Pa. to Ill. and Mont., also Calif. and Ore. (4). II and III on Carex spp.
P. fraseri Arth. (III, ?II). Pa., Tenn., Va., W.Va. (3); Mont., Nebr., N.H., N.Y., Pa. (4)
P. hieracii (Schum.) Mart. (0,I,II,III). Tex. (1); Me. to Va., Ill., and Wash. (2); N.Y., N.Car., Pa. (3); on numerous other spp. in this range, also Fla., Calif., Colo., and Alaska.
P. maculosa Schw. (III). Ore., Tex. Wash. (4)
Septoria cercosperma Rostr., leaf spot. Tex. (1,4).
S. hieracicola Dearn. & House. N.Y. (4)
Sphaerotheca humuli (DC.) Burr. var. fuliginea (Schlecht.) Salm., powdery mildew. Ill. (4)

HYMENOPAPPUS (COMPOSITAE)

HYMENOPAPPUS CAROLINENSIS (Lam.) Porter. Biennial herb of the Southeastern and South Central States, sometimes grown in wild gardens and useful to wildlife.

Phymatotrichum omnivorum (Shear) Dug., root rot. Tex.
Plasmopara halstedii (Farl.) Berl. & Det., downy mildew. Okla.
Puccinia grindeliae Pk. (0,III), rust. Okla.

HYPOCHOERIS (COMPOSITAE)

HYPOCHOERIS GLABRA L. (1) and H. RADICATA L. (2) CATS-EAR, FALSE-DANDELION. Annual (1) or perennial (2) herbs of Europe, naturalized in the Pacific States; (2) especially a troublesome weed but useful to wildlife.

Ditylenchus dipsaci (Kuehn) Filip., stem nematode. Calif., Ore., Wash., Hawaii (2)

HYPOCHOERIS cont.

Phymatotrichum omnivorum (Shear) Dug., root rot. Tex. (2)
Puccinia hieracii (Schum.) Mart. (0,I,II,III), rust. Calif. (1,2), Ore. (2)

INULA (COMPOSITAE)

INULA HELENIUM L., ELECAMPANE. Coarse perennial of Europe and Asia, naturalized in the Eastern States; sometimes grown for ornament and for roots, a source of inulin, also useful to wildlife.

Anthostomella rostrospora (Gerard) Sacc., on stems. N.Y.
Erysiphe cichoracearum DC., powdery mildew. N.Y., Wis.
?Puccinia hieracii (Schum.) Mart. (II,III), rust. Wis.
Ramularia sp., leaf spot. Mich.

KRIGIA (COMPOSITAE)

KRIGIA BIFLORA (Walt.) Blake, DWARF-DANDELION (1); other spp. (2). Annual or perennial herbs of the Eastern and Central States, sometimes grown for ornament and useful to wildlife.

Bremia lactucae Regel, downy mildew. Wis. (1,2). Miss., Mo., Okla. (2)
Mycosphaerella krigiae (Ell. & Ev.) Greene, leaf spot. Ill., Wis. (1)
Phoma krigiae Tassi, on stems and bracts. (2)
Phymatotrichum omnivorum (Shear) Dug., root rot. Tex. (2)
Puccinia extensicola Plowr. var. hieraciata (Schw.) Arth. (0,I), rust. Ill., Iowa, Wis. (1). II and III on Carex spp.
P. hieracii (Schum.) Mart. (0,I,II,III). N. Car. (1)
P. maculosa Schw. (III). Ill., Mich., Mo., Pa., Tenn. (1); Miss., Mo., Pa. (2).
Septoria krigiae Dearn. & House, leaf spot. Ky., N.Y., Wis. (1)

KUHNIA (COMPOSITAE)

KUHNIA EUPATORIOIDES L., FLASE BONESET. Perennial herb of the Eastern and Central States and southward, sometimes grown in wild gardens. Other spp. (2)

Phymatotrichum omnivorum (Shear) Dug., root rot. Tex. (2)
Pleospora compositarum Earle, leaf spot. N.Mex., Tex. (2)
Puccinia kuhniae Schw. (0,I,II,III), rust. Ind. to Ala., Nebr., and N.Dak. (1); on other spp. to Fla., Tex., and Mont.

LACTUCA (COMPOSITAE)

LACTUCA SATIVA L., LETTUCE. Cultivated annual, perhaps derived originally from L. serriola L., commonly grown in gardens as a salad plant; commercial cultivation predominates in Calif. and Ariz.; other leading states: Colo., Fla., Ida., N.J., N.Y., Ore., Wash. In general these records apply alike to all the horticultural types, such as var. CAPITATA L., HEAD L., var. CRISPA L., LEAF L., and var. LONGIFOLIA Lam., COS or ROMAINE L.

Albugo sp., white rust. Tex.

Alternaria sp., secondary leaf spot. Fla., La., Ohio, Okla., Tex., Wash.; occasional in northeastern States.

Botrytis cinerea Pers. ex Fr., gray mold rot (chiefly in greenhouses and in markets, sometimes in fields, especially on seed plants). Cosmopolitan. A stem girdle of growing plants is also reported in New York.

Bremia lactucae Regel, downy mildew. General.

Cercospora longissima Sacc., leaf spot. Fla., Ill., Ind., Tex., Va., Wis. P.R.

Erwinia carotovora (L.R.Jones) Holland, bacterial soft rot, slime. Cosmopolitan in transit and markets.

Erysiphe cichoracearum DC., powdery mildew. Calif., Mich.

Fusarium sp., root rot, wilt. Ky., Ohio.

Heterodera marioni (Cornu) Goodey, root knot. General from Va. to Fla. and Ariz.; Hawaii; occasional in greenhouses in the North.

Marssonina panattoniana (Berl.) Magn., anthracnose. N.Y. to Fla., Tex. and Mich.; Calif., Ore., Wash.

Phymatotrichum omnivorum (Shear) Dug., root rot. Ariz., Tex.

Physarum cinereum Batsch ex Fr., slime mold. Occasional in hotbeds and greenhouses.

Pleospora herbarum (Pers. ex Fr.) Rab. (Stemphylium botryosum Wallr.), seed mold, secondary leaf spot. Fla., Ky., N.Y.

Pseudomonas marginalis (N.A.Brown) F.L.Stevens, marginal leaf blight. Kans., Mo., N.J., N.Y.

P. rhizoctonia (R. C. Thomas) Burkh., bacterial rosette. N.Y., Ohio, Pa.

P. viridilivida (N.A.Brown) Holland, bacterial rot. La., Va., Wash.; ? Del., N.H.

Puccinia extensicola Plowr. var. hieraciata (Schw.) Arth. (O.I), rust. Ind., Minn., N.Dak., Wis. II and III on Carex spp.

P. hieracii (Schum.) Mart. (II), rust. Calif. Other stages not observed.

Pythium spp., damping off, root rot, stunt, wilt. Cosmopolitan. Commonly identified as P. debaryanum Hesse but frequently is P. ultimum Trow; P. polymastum Drechs. is reported in Conn. The specific cause of a form of root rot termed stunt, as reported in Mich., Mo., and N.Y., has not been identified.

Rhizoctonia solani Kuehn (=Pellicularia filamentosa (Pat.) Rogers),

LACTUCA cont.

bottom rot, rosette, damping off. General. The basidial stage occasionally develops on this host.

Sclerotinia sclerotiorum (Lib.) DBy., drop, watery soft rot. Widespread in greenhouse and field culture, also in transit and markets. S. minor Jagger, causing a similar disease, is reported in Fla., La., Mass., N.J., NY., Ore., Pa.

Sclerotium rolfsii Sacc., southern blight. Calif., Fla., N.&S. Car., Tex., Va.

Septoria lactucae Pass., leaf spot. Occasional in the Eastern and Central States to Fla., Colo. and Minn.; Hawaii

Xanthomonas vitians (N.A.Brown) Starr & Weiss, bacterial wilt. N.J., N.Mex., N.Y., Pa., S.Car., Va., ? Hawaii.

Big vein -- unidentified virus. Ariz., Calif., Colo., Ill., Md., N.J., N.Y.

Mosaic -- virus (Marmor lactucae Holmes, Lactuca virus 1 K.M.Sm.). Widespread, probably general.

Spotted wilt -- virus (Lethum australiense Holmes, Lycopersicon virus 3 K.M.Sm.). Calif., Tex.

Yellows (white heart, Rio Grande disease) -- virus (Chlorogenus callistephi Holmes, Callistephus virus 1 K.M.Sm.) Northeastern and Central States to Miss. and Tex.; Colo., Ida., Ore., Wyo. Also var. californicus Holmes, Callistephus virus 1A K.M.Sm. in Calif.

Brown blight -- cause unknown. Ariz., Calif., sometimes reported in markets on produce from these areas. A disease ascribed to an unidentified virus is reported under this name in Ill. and Ind.

Tipburn -- physiological, attributed to excessive transpiration and insolation or to high temperature. General.

LACTUCA spp. ("wild"), as L. CANADENSIS L., CANADA WILD LETTUCE (1); L. GRAMINIFOLIA Michx., GRASSLEAF L. (2); L. PULCHELLA (Pursh) DC., CHICORY L. (3); L. SERRIOLA L., including forma INTEGRIFOLIA Bogenh., PRICKLY L. (4); L. SPICATA (Lam.) Hitchc., BLUE L. (5); other spp. (6). Biennial or perennial herbs of plains, woodlands, and wasteground, native in the Eastern and Central States (1,2,3,5) or European but widely naturalized in the U. S. (4). Several are useful to wildlife; (4) is a troublesome weed; its var. sativa CELTUCE is grown for edible stems.

Alternaria sonchi J.J.Davis, leaf spot. Ill. (1); Tex. (1,4,6)

Asteroma lactucae J.J.Davis, on leaves. Tex., Wis. (1,6)

Bremia lactucae Regel, downy mildew. Reported on all spp. listed except (2), also on others, throughout their respective ranges from Mass. to Miss., Tex., Calif., and Wash.

Calosphaeria herbicola Ell. & Ev., leaf spot. Tex. (1,4,6)

LACTUCA cont.

Cercospora longissima Sacc., leaf spot. La. (6)
Cuscuta gronovii Willd., dodder. N.Y. (4)
Cylindrosporium lactucicola Ell. & Ev., N.Car. (4); Colo. (6)
Diaporthe arctii (Lasch) Nits., on stems. Ga. (1); La. (6)
Ditylenchus dipsaci (Kuehn) Filip., stem nematode. N.Y. (1)
Entyloma compositarum Farl., leaf smut. Minn. (1); Tex. (1,4,6)
Erysiphe cichoracearum DC., powdery mildew. Reported on all spp. named except (2), and on others, within their ranges from Mass. to N.Car., N.Mex., Calif., and Mont.
Gibberidea heliopsidis (Schw.) Shear, black patch. Conn. (6)
Heterodera marioni (Cornu) Goodey, root knot.
Leptothyrium lactucae Dearn. & Barth., on leaves. Kans. (4)
Marssonina panattoniana (Berl.) Magn., anthracnose. Ida., Wash. (4)
Mycosphaerella depressa (Pk.) House, on stems. Kans., N.Y. (1); Mich. (6). An invalid name.
Ovularia carletonii Ell. & Kell., leaf spot. Colo., Kans., Nebr., S.Dak. (3); Kans. (6).
Phyllosticta spp., leaf spot. P. decidua Ell. & Kell., Wis. (1); P. lactucae Atk., Ala. (1); Tex. (1,4); P. mulgedii J.J.Davis, Wis. (5); P. scariolicola Tehon, Ill. (4).
Phymatotrichum omnivorum (Shear) Dug., root rot. Tex. (4)
Plasmopara halstedii (Farl.) Berl. & DeT., downy mildew. Md. (1)
Puccinia extensicola Plowr. var. hieraciata (Schw.) Arth. (0,I), rust. Widespread on all spp. named, and others, in their respective ranges from Mass. to Ill., Colo. and Ida. II and III on Carex spp.
P. minussensis Thuem. (0,I,II,III), rust. Mich., N.Y. (1); Wis. to N.Mex., Utah, and Wash. (3).
Rhabdospora solidaginis Cke. & Ell., on stems. Colo. (6)
Rhizoctonia solani Kuehn, root and stem rot. N.Y. (4)
Sclerotinia sclerotiorum (Lib.) DBy., stem rot. Calif., ?Mass. (4); Md., Miss. (6)
Sclerotium bataticola Taub., charcoal rot. Ill. (4)
S. rolfsii Sacc., southern blight. Tex. (1,4,6)
Septoria lactucae Pass., leaf spot. Central and Eastern States (1,4,6), also Calif., Ga., La. (4); Fla. (2); Wis. (3); Ala. (5)
S. lactucicola Ell. & Mart., leaf spot. N.Y. to Miss., Okla., and Wis. (1); Mich., Ohio, Wis. (4); Me. to Fla., Colo., and S.Dak. (6)
S. unicolor Wint., leaf spot. Kans. (1); Tex. (1,4); Colo., Ill., Mo., Tex. (6)
Sphaerotheca humuli (DC.) Burr., var. fuliginea (Schlecht.) Salm., powdery mildew. Md., Wis. (1); Mich. (5,6); Ala., Ind., Md., N.J., Wis. (6)
Synchytrium aureum Schroet., leaf gall. La. (6)
Xanthomonas lactucae-scariolae (Thornberry & H.W.Anders.) Burkh., bacterial leaf spot. Ill. (4)

LACTUCA cont.

Spotted wilt -- virus (Lethum australiense Holmes, Lycopersicon virus 3 K.M.Sm.). Tex. (1,2)

Yellows -- virus (Chlorogenus callistephi Holmes, Callistephus virus 1 K.M.Sm.). Tex. (1,4), Kans. (4,6), also var. CALIFORNICUS in Calif. (4)

LEONTODON (COMPOSITAE)

LEONTODON AUTUMNALIS L., FALL-DANDELION, HAWKBIT. Perennial herb of Europe, sometimes grown in gardens and naturalized in the Northeast; useful to wildlife.

Puccinia hieracii (Schum.) Mart. (0,I,II,III), rust. Me., N.H.

Yellows -- virus (Chlorogenus callistephi Holmes, Callistephus virus 1 K.M.Sm.). Me., N.Y.

LIATRIS (COMPOSITAE)

LIATRIS spp., GAY-FEATHER. Erect perennial herbs of prairies and open ground in the Eastern and Central States, also west and south; often grown for ornament. Those most commonly cultivated are L. PYCNSTACHYA Michx. (1), L. SCARIOSA Willd. (2), L. SPICATA Willd. (3); other spp. (4)

Coleosporium laciniariae Arth. (II,III), rust. Tex. (1); Ark., Fla., Ga., Tenn. (2); N.J. to Fla. and Ark. (4). 0 and I on 2- and 3-needle Pinus spp.

Cuscuta glomerata Choisy, dodder. Okla. (4)

Erysiphe cichoracearum DC., powdery mildew. Okla. (2)

Helminthosporium macrocarpum Grev. Mich. (2)

Heterodera marioni (Cornu) Goodey, root knot.

Phyllosticta liatridis J. J. Davis, leaf spot. Wis. (3)

Phymatotrichum omnivorum (Shear) Dug., root rot. Tex. (1)

Puccinia liatridis [Webber] Bethel (0,I), rust. Ind. (1); Wis. (1,3); Kans., N.Dak. (2); Nebr. (3); Wis. to Colo. and Mont. (4). II and III on Koeleria cristata (L.) Pers. and Agrostis spp.

Sclerotinia sclerotiorum (Lib.) DBy., stem rot. N.Y. (4)

Septoria liatridis Ell. & J.J.Davis, leaf spot. Tex. (1); Wis. (1,2,3,4); N.Dak. (2); Minn. (4)

Verticillium albo-atrum Reinke & Berth., wilt. N.J. (1,2,4)

MALACOTHRIX (COMPOSITAE)

MALACOTHRIX ARACHNOIDEA McGregor (1), other spp. (2). Monthly annual herbs of plains and deserts in the far West; sometimes grown for ornament.

Puccinia harknessii Vize (III), rust. Calif. (1,2)
P. hieracii (Schum.) Mart. (0,I,II,III), rust. Calif. (2)
Synchytrium innominatum Farl., leaf gall. Calif. (2)

MATRICARIA (COMPOSITAE)

MATRICARIA CHAMOMILLA L., FALSE-CHAMOMILE (1), and M. MATRICARIOIDES (Less.) Porter, PINEAPPLE-WEED (2). Annual herbs, (1) of Europe, sometimes grown in herb gardens and escaped in the Eastern States; (2) native of the far West, extensively naturalized, an occasional weed.

Albugo tragopogonis Pers. ex S.F.Gray, white rust. Calif., N.Dak., Ore., Wash. (2)
Erysiphe cichoracearum DC., powdery mildew. Wash. (2)
Heterodera marioni (Cornu) Goodey, root knot. (1)
Sphaerotheca humuli (DC.) Burr. var. fuliginea (Schlecht.) Salm., powdery mildew. Wash. (2)

Yellows -- virus (Chlorogenus callistephi var. californicus Holmes, CALLISTEPHUS virus 1A K.M.Sm.) Calif. (2)

MIKANIA (COMPOSITAE)

MIKANIA SCANDENS (L.) Willd., CLIMBING HEMPWEED (1). Twining herb of the Eastern and Southern States and southward, sometimes grown in wild gardens and for plant cover. Other spp. (2).

Cercospora mikaniae Ell. & Ev., leaf spot. Miss. (1)
C. mikanicola F.L.Stevens. P.R. (2)
Endophylloides portoricensis Whet. & Olive (III), rust. Canal Zone, P.R. (2)
Irene sororcula (Speg.) F.L.Stevens, black mildew. P.R. (2)
Meliola compositarum Earle, black mildew. P.R. (2)
Puccinia spegazzinii DeT. (III), rust. Ala., Fla., Miss., N.Car., Canal Zone, P.R. (1); Fla., P.R. (2)
Septoria mikanii Wint., leaf spot. Conn. Tex. (1); P.R. (2)

PARTHENIUM (COMPOSITAE)

PARTHENIUM ARGENTATUM A. Gray, GUAYULE (1). Low shrub of Mexico and southwestern U. S., cult. commercially in California as source of rubber. P. INTEGRIFOLIUM L., AMERICAN FEVERFEW (2). Perennial herb of the Eastern and Central States. Other spp. (3)

Albugo tragopogonis Pers. ex S.F.Gray, white rust. Iowa (2), Kans. (3)
Alternaria sp., secondary leaf spot. Calif. (1)
Botrytis cinerea Pers. ex Fr., seedling blight. gray-mold rot. Calif. (1)
Cercospora partheniphila Chupp & Greene, leaf spot. Wis. (2,3)
Coleosporium terebinthinaceae (Schw.) Arth. (II,III), rust. Ala., Ga., La., N.Car., Tenn., Va. (1); Ga., N.Car., Tenn., Va. (3). O and I on 2- and 3-needle Pinus spp.
Diplodia theobromae (Pat.) Nowell, dieback. Calif., Tex. (1)
Erwinia carotovora (L.R.Jones) Holland f. parthenii Starr, bacterial stem rot. Calif. (1)
Heterodera marioni (Cornu) Goodey, root knot (resistant). Ariz. (1)
Macrophomina phaseoli (Maub.) Ashby, charcoal rot. Calif., Tex. (1)
Phymatotrichum omnivorum (Shear) Dug., root rot. Ariz. (1); Tex. (1,3).
Phytophthora drechsleri Tucker, root rot. Ariz., Calif., N.Mex., Tex. (1)
Puccinia parthenii (Spég.) Arth. (II,III), rust. Tex. (3). O and I unknown
P. melampodii Diet. & Holw. (III). Tex. (3)
Pythium ultimum Trow, damping off, pink root rot. Calif. (1)
Rhizoctonia solani Kuehn, damping off. Calif. (1)
Sclerotinia minor Jagger, and S. sclerotiorum (Lib.) DBy., stem rot. Calif. (1)
Sclerotium rolfsii Sacc., southern blight. Ariz. (1)
Verticillium albo-atrum Reinke & Berth., wilt. Ariz., Calif., N.Mex., Tex. (1)

Yellows -- virus (Chlorogenus callistephi Holmes, Callistephus virus 1 K.M.Sm.). Tex. (3)

PETASITES (COMPOSITAE)

PETASITES FRIGIDUS (L.) Fr. (1), and P. PALMATUS (Ait.) A. Gray (2), BUTTER-BUR. Perennial herbs of northern latitudes, sometimes grown in wild gardens (European spp. more commonly than these).

Puccinia poarum Niels. (O,I), rust. Alaska (1). II and III on Poa and Festuca in Europe.

PETASITES cont.

P. conglomerata (Strauss) Schmidt & Kunze (III). Alaska (1); Mich., Minn., N.Y., Wis. (2)
Ramularia variegata Ell. & Holw., leaf spot. Wis. (2)
Stagonospora petasitidis Ell. & Ev., leaf spot. Wis. (2)
Synchytrium aureum Schroet., leaf gall. Wis. (2)

PIQUERIA (COMPOSITAE)

PIQUERIA TRINERVIA Cav. (Stevia serrata Hort.), STEVIA. Perennial herb of Mexico and Central America, grown under glass and widely used as filling material in floriculture.

Rhizoctonia solani Kuehn, damping off. Ill.

Yellows -- virus (Chlorogenus callistephi Holmes, Callistephus virus 1 K.M.Sm.). Mich., N.J.

PLUCHEA (COMPOSITAE)

PLUCHEA CAMPHORATA (L.) DC., MARSH FLEABANE (1). Coarse annual of salt marshes of the Atlantic and Gulf Coasts, useful to wildlife. Other spp. (2).

Heterodera marioni (Cornu) Goodey, root knot. ? Fla. (2)
Phleospora plucheae Stevenson, on leaves. Tex. (2)
Puccinia plucheae (Syd.) Arth. (I,II,III), rust. Fla. (1); P.R. (2)
Septoria plucheae Guba leaf spot. Mass. (1)

PRENANTHES (COMPOSITAE)

PRENANTHES spp., RATTLESNAKE-ROOT, as P. ALBA L. (1), P. ASPERA Michx. (2), P. TRIFOLIOLATA (Cass) Fern. (3), other spp. (4). Perennial herbs of woodlands (1,3) and plains (2) in the Eastern and Central States, sometimes grown in wild gardens.

Bremia lactucae Regel, downy mildew. Iowa, Mass., Minn. (1); Mass. (4)
Cercospora brunnea Pk., leaf spot. Wis. (1); Ark., N.Car. (4)
C. prenanthis Ell. & Kell. Ind. (1); Kans. (2); Ala. (4)
C. tabacina Ell. & Ev. Wis. (1)
Diplodia natalensis Pole-Evans on stems. Ala. (4)
Erysiphe cichoracearum DC., powdery mildew. N.Y. (1); Ill., Md., N.Car., Pa. (4)

PRENANTHES cont.

Laestadia prenanthis Ell. & Ev., leaf spot. Ala. (4)
Phlyctaena simulans (Berk. & Curt.) Sacc., on stems. N.H. (4)
Puccinia atropuncta (0,I), rust. Pa., Va. (3). II and III on Amianthium muscaetoxicum.
P. extensicola Plowr. var. hieraciata (Schw.) Arth. (0,I). Ill., Minn., N.Y., Wis. (1,4); Va. (3). II and III on Carex spp.
P. insperata Jacks. (I,II,III). Ore., Alaska (4)
P. orbicula Pk. & G.W.Clint. (0,I,II,III). Me. to Tenn. and N.Dak. (1 and 3 within their ranges, 4).
Septoria nabali Berk. & Curt., leaf spot. N.Y. to Iowa and Wis. (1); Va. (3); Me. to Va., Ohio, and Wis. (4)
Sphaerotheca humuli (DC.) Burr. var. humuli (Schlecht.) Salm., powdery mildew. Ohio, Pa., Wis. (1); N.Y. to Ala., Ill. and Minn.; Alaska (4).
Synchytrium aureum Schroet., leaf gall. Wis. (1)

RATIBIDA (LEPACHYS) (COMPOSITAE)

RATIBIDA COLUMNARIS (Sims) D. Don (1) and R. PINNATA (Vent.) Barnhart (2), prairie coneflower. Perennial herbs of dry prairies in the Central States and westward (1), east and south (2); sometimes grown in gardens, especially (1).

Cercospora ratibida Ell. & Barth., leaf spot. Kans. (1); Wis. (2)
Entyloma compositarum Farl. (E. lepachydis Cif.), white smut. Nebr. (1); Mich. to Ind., Kans., and Minn. (2)
Erysiphe cichoracearum DC., powdery mildew. N.Dak., Tex. (1)
Plasmopara halstedii (Farl.) Berl. & DeT., downy mildew. Iowa (2)
Phymatotrichum omnivorum (Shear) Dug., root rot. Tex. (1)
Physalospora lepachydis Ell. & Ev., leaf spot. Mont. (1)
Ramularia rudbeckiae Pk., leaf spot. Ida. (1)
Rhizoctonia crocorum (Pers. ex DC.) Fr., violet root rot. Tex. (1)
Septoria infuscata Wint., leaf spot. Mich., Mo., Wis. (2)
S. lepachydis Ell. & Ev. Kans. (1); Wis. (2)
S. rudbeckiae Ell. & Halst. N.Mex., N.Dak. (1)
Uromyces perigynius Halst. (0, I), rust. N.Dak., Tex. (1)

RUDBECKIA (COMPOSITAE)

RUDBECKIA HIRTA L., BLACK-EYED SUSAN (1); R. LACINIATA L., especially the cultivated double form, var. HORTENSIA Bailey, GOLDEN-GLOW (2); R. TRILOBA L., BROWN-EYED SUSAN (3); other spp. (4). Biennial or sometimes annual (1,3), or perennial (2), herbs, widely distributed in nature and grown in gardens.

Aecidium batesii Arth. (0,I), rust. Nebr. (1)
Cercospora rudbeckiae Pk., leaf spot. N.Y. (2)

RUDBECKIA cont.

C. tabacina Ell. & Ev. Wis. (1,2); Ill., N.Y. (2)
Colletotrichum rudbeckiae Pk., on stems. N.Y. (2)
Diaporthe arctii (Lasch) Nits., on stems. Ga. (2)
Entyloma compositarum Farl. (E. anceps Cif.), white smut. Iowa, Mo., Ohio, Wis. (2); Iowa (4)
E. polysporum (Pk.) Farl. (E. davisii Cif.). Wis. (1)
Erysiphe cichoracearum DC., powdery mildew. Mass., S.Dak., Va. (1); general (2); Utah (4)
Mycosphaerella rudbeckiae Clements, on stems. Colo. (1)
Phyllosticta rudbeckiae Ell. & Ev., leaf spot. Iowa, N.Y., Wis. (2)
Phymatotrichum omnivorum (Shear) Dug., root rot. Tex. (4)
Plasmopara halstedii (Farl.) Berl. & DeT., downy mildew. N.Y., N.Dak. (1); Iowa, Nebr., N.Car., N.Dak., Wis. (2)
Puccinia extensicola Plowr. [? var. rudbeckiae Arth.] (0,I), rust. Md., S.Dak. (2). Has been so reported but identity with this sp. having II and III on Carex has not been proved by cultures; perhaps Uromyces perigynius.
P. rudbeckiae Barth. (III). Tex. (4)
Ramularia rudbeckiae Pk., leaf spot. Vt. to Miss., Colo., and Ida. (2); Mont., Va., Vt., W.Va. (4)
Sclerotinia sclerotiorum (Lib.) DBy., stem rot. Conn. (2)
Sclerotium rolfsii Sacc., southern blight. Fla. (2); N.J. (4)
Septoria rudbeckiae Ell. & Halst., leaf spot. Del., Wis. (1); Kans., Nebr., Wis. (2); Colo., Ill., Mass., Miss., Wash. (4)
var. oaklandica Sacc. N.Y. (1)
Synchytrium aureum Schreot., leaf gall. Ill., Wis. (2)
Uromyces perigynius Halst. (0,I), rust. Md. to Mo. and Mont. (2); II and III on Carex spp.
U. rudbeckiae Arth. & Holw. (III). ? Mont. (1); Md. to Miss., N. Mex., and Mont. (2); Miss. (3); Tex. (4)
Verticillium albo-atrum Reinke & Berth., wilt. N.Y. (1)

Mosaic -- unidentified virus. Ind. (2) Ill. (4)
Yellow dwarf -- virus (Aureogenus vastens (Holmes) Black, Solanum virus 16 K.M.Sm. N.Y. (1)
Yellows -- virus (Chlorogenus callistephi Holmes, Callistephus virus 1 K.M.Sm.). N.Y. Also var. californicus Holmes, in Calif.

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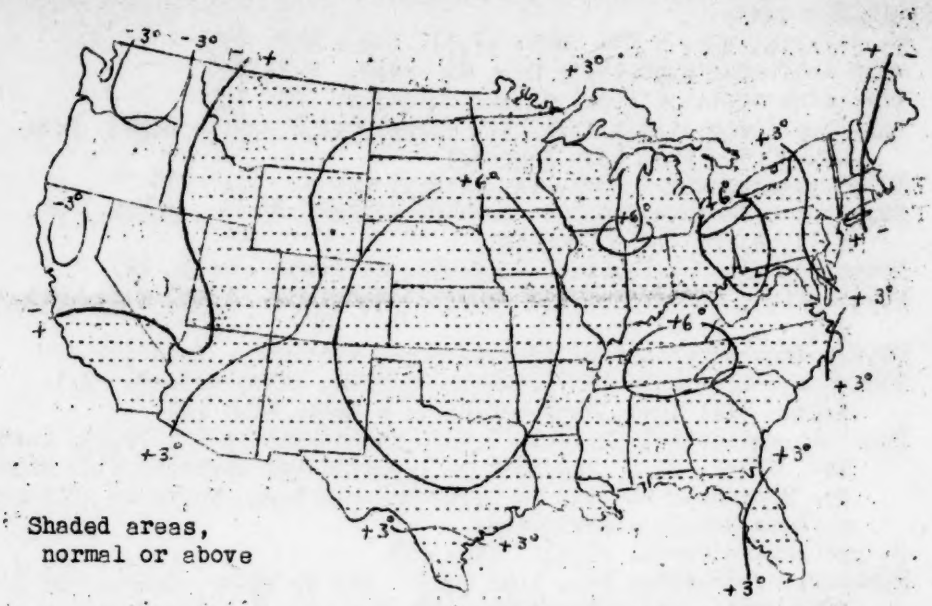
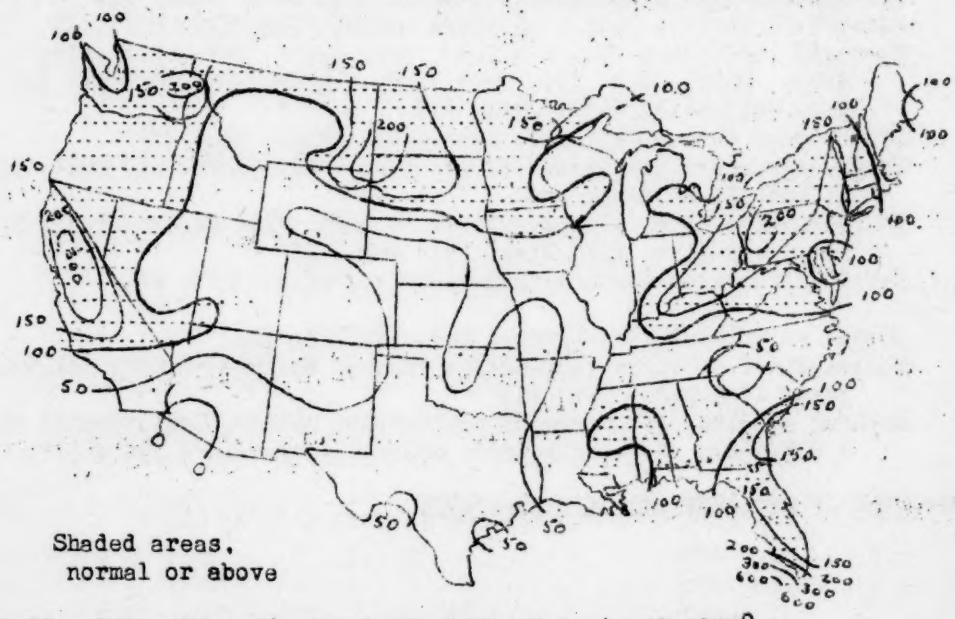


Map I. Departure of mean temperature from normal, April 1948.



Map II. Percentage of normal precipitation, April, 1948

APRIL WEATHER

From U. S. Department of Commerce, Weather Bureau, Weekly Weather and Crop Bulletin for week ending May 4, 1948.